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SAMPLING AND ANALYSIS PLAN FOR THE DETERMINATION OF THE NATURE AND EFFECTS OF HEAVY METALS WITHIN THE WETLAND AND POND AREAS AT RICHARDSON FLAT

SITE ID: UT980952840

Richardson Flat Site
Park City, Utah

PART I – FIELD SAMPLING PLAN
PART II – QUALITY ASSURANCE PROJECT PLAN

March 14, 2003

Prepared by:

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Chris Cline	

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LIST OF ACRONYMS AND ABBREVIATIONS

AVS	Acid Volatile Sulfide
CLP	Contract Laboratory Program
COC	Contaminant of Concern
DO	Dissolved Oxygen
DOC	Dissolved Organic Carbon
DQO	Data Quality Objective
EDD	Electronic Data Deliverable
EPA	U.S. Environmental Protection Agency
FSP	Field Sampling Plan
FTP	Field Technical Procedure
GPS	Global Positioning System
HASP	Health and Safety Plan
ICP	Inductively Coupled Plasma
ICP/MS	Inductively Coupled Plasma Mass Spectrometry
IDW	Investigation Derived Waste
LCS/LCSD	Laboratory Control Sample/Laboratory Control Sample Duplicate
MOU	Memorandum of Understanding
MS/MSD	Matrix Spike/Matrix Spike Duplicate
NPL	National Priorities List
PARCC	Precision, Accuracy, Representativeness, Completeness, and Comparability
PPE	Personal Protective Equipment
PRG	Preliminary Remediation Goal
QA	Quality Assurance
QC	Quality Control
QAPP	Quality Assurance Project Plan
QMP	Quality Management Plan
QP	Quality Procedure
RPD	Relative Percent Difference
RPM	Remedial Project Manager
TERA	Terrestrial Ecological Risk Assessment
SAP	Sampling and Analysis Plan
SEM	Simultaneously Extracted Metals
SOP	Standard Operating Procedure
SPLP	Synthetic Precipitation Leaching Procedure
TAL	Target Analyte List
TOC	Total Organic Carbon
TOM	Total Organic Matter
UCL	Upper Confidence Level

USFWS	U.S. Fish & Wildlife Service
%R	Percent Recovery

1.0 INTRODUCTION

This document serves as the Sampling and Analysis Plan (SAP) for surface water, sediment, pore water, benthic macroinvertebrate, fish, and vegetation sampling, and toxicity testing of invertebrate species exposed to sediments collected from a wetland and a pond located adjacent to the Richardson Flat Site tailings impoundment. This work is being conducted as part of the Focused Remedial Investigation Feasibility Study (RIFS) that United Park City Mines Company (United Park) voluntarily agreed to conduct in the Administrative Order on Consent dated September 28, 2000.

There have been numerous site investigations conducted at this site over the past seventeen years by the Environmental Protection Agency (EPA), Utah Division of Environmental Response & Remediation (DERR) and United Park. There have been significant remedial activities performed by United Park over the past ten years including capping the surface of the tailings with clay soils and reconstruction of the south diversion ditch. These studies and remedial activities are summarized in the Remedial Investigation SAP (RMC, 2001).

In January of 2001 EPA formed a Biological Technical Assistance Group (BTAG) to ensure that the appropriate stakeholders were represented in evaluating ecological risks presented by the site. Members of the BTAG are EPA, United Park, DERR and the United States Fish & Wildlife Service (USFWS). Members of the BTAG group reviewed and commented on the Remedial Investigation SAP that was initiated in April of 2001. In August of 2001 the group met to review EPA's draft of the Screening Ecological Risk Assessment (SERA) prepared by Syracuse Research Corporation (Syracuse, 2001), a revised draft was submitted to the group in March of 2002. This SAP has been prepared to address data gaps identified in the recent Draft SERA, March 2002.

This SAP will address data gaps identified in the recent SERA for the wetland and pond located in the northwestern portion of the site, adjacent to the main embankment. Data gaps for the on and off impoundment soils will not be addressed in this SAP, future remedial activities planned by United Park will address potential ecological risks in the soils. If United Park does not carry out the remedial activities then the data gaps identified in the SERA will be completed and United Park will conduct an ecological risk assessment for those areas.

Figure 1.0 shows the geographic location of the site.

The SAP is comprised of the Field Sampling Plan (FSP) and the Quality Assurance Project Plan (Plan) and includes the following sections:

Section 1 -Introduction
Section 2 -Site Background

Part I: Field Sampling Plan

Section 3 - Sampling Program, Rationale, and Locations
Section 4 – Field Activity Methods and Procedures

Part II: Quality Assurance Project Plan

Section 5 - Project Management
Section 6 – Quality Control Requirements
Section 7 - Assessment and Oversight
Section 8 - Data Validation and Usability
Section 9 - Measurement and Data
Section 10 - Acquisition References

Appendix A - Standard Operating Procedures
Appendix B – AEC QA/QC Documentation

1.1 Objectives

This SAP describes the collection and analyses of surface water samples, sediment (0–10 cm depth) and pore water samples, and organism tissue samples, as well as observations on benthic macroinvertebrate and plant communities. The samples will be collected from the wetland area west of the main embankment and the pond area located near the terminus of the diversion ditch (Figure 1.1). The sampling effort will provide data to determine:

- Potential and actual risk to terrestrial and aquatic receptors from exposures to heavy metals in the wetland and pond surface water, sediments, pore waters, and biota;
- If unacceptable risk levels are found, the data will be used to determine preliminary remediation goals (PRGs) for the affected areas.

Figure 1.1 shows the location of the wetland and the pond where potential metals contamination and risks will be evaluated. Metals levels in the wetland and pond sediments may present unacceptable risks to native vegetation, invertebrates, and fish, as well as wildlife. To determine if unacceptable risks exist, RMC, as advocated by the BTAG, will focus this investigation on the bioavailable fraction of metals in the sediments and their relationship with *in situ* plant and benthic macroinvertebrate community indices; plant, invertebrate, and fish tissue burdens; and laboratory sediment toxicity tests. Furthermore, sediment quality parameters such as grain size distribution, pH, moisture, total organic carbon, and nutrients will be quantified to develop predictive relationships between bulk sediment concentrations and bioavailable metals (Suave et al. 1998). These relationships will then be used in the calculation of bulk (or total) sediment metal concentrations used as PRGs, if needed. In addition, sediment pore water samples will be collected to determine the relationship between bulk metal concentrations and pore water concentrations. This will aid in determining fate and transport of metals in the wetland and pond sediments.

Data collected as part of this SAP will build upon the existing Draft SERA. Default values used in the SERA resulted in an overestimate of risk. Site-specific data collected as part of this SAP will be compared to appropriate reference site data and used to more accurately assess ecological risks at the site and to address data gaps identified in the Draft SERA (Syracuse, 2002).

1.2 Project Schedule and Deliverables

Data collection will occur following EPA approval of this SAP in late spring to early summer. Timing of the data collection will be dependent upon snowmelt in the contributing watershed, emergence of sufficient vegetation for sample collection, and presence of sufficient numbers of aquatic macroinvertebrates for tissue collection and community analysis. Sediment, pore water, surface water, and macroinvertebrate samples for community analysis will be collected synoptically. Collection of tissue samples and the plant community observations may be done separately from other sampling depending on the availability of organisms. Deliverables will include a description of sampling activities and data when available from the laboratories in the RIFS monthly reports, and a final report describing sampling activities and summarizing the data for EPA use in the ecological risk assessment.

2.0 SITE BACKGROUND

A detailed description of the Site, including a description of the Site operational history, existing closure measures and elements, regional geology and hydrogeology and surface water is set forth in Sections 2.0 to 2.5 of the Focused RI/FS Work Plan (RMC, 2000). The study area, site history, previous site investigations, and environmental setting are summarized below.

2.1 Study Area

United Park is the current owner of a large parcel of property (the "Property"), comprising approximately 700 acres, located in Summit County, Utah (Figure 2.0). The Site included a historic mine tailings impoundment consisting of a 160-acre, geometrically closed basin formed by an earth embankment and a series of perimeter containment dikes. The tailings impoundment resulted from decades of mining and milling silver-laden ore in the area around Park City known as the Park City Mining District. The tailings impoundment is now covered by clean soils. A wetland area on the site encompasses approximately eight (8) acres, with a pond just south of the wetland is approximately one (1) acre in surface area. Water exiting from the pond along the embankment on the south side of the tailings impoundment flows in a southeasterly to northwesterly direction in a discrete channel where it mixes with a portion of the Silver Creek flow in the northwestern corner of the wetland area.

Silver Creek forms the western boundary of the site. Several man made and natural barriers affect the flow pattern of Silver Creek near the site. Near the northwestern corner of the wetland area, Silver Creek flows into the wetland beneath the rail trail bridge where it is split by a topographical high. At that point, a portion of the flow travels to the east and mixes with the flow from the diversion ditch and a portion moves to the northeast where it converges with the wetland drainage and exits the wetland area in a concrete box culvert under State Highway 248.

2.2 Site History

The Site has remained unused since mining and milling operations ceased in 1982. Land use in the Study Area is presently limited to onsite workers, site investigation activities, and wildlife habitat. In the wetland and pond areas, current and future land use will be limited to wildlife habitat.

Over the past fifteen years, the (EPA), the Utah Department of Environmental Quality (UDEQ) and United Park have been investigating the Site in order to characterize the Site and determine potential adverse impacts to human health and the environment associated with the Site. At the same time, United Park has been implementing a series of remedial measures at the Site intended to mitigate any potential adverse impacts on human health and the environment.

Remedial measures conducted by United Park include the following:

- Placement of clean cover soils over all exposed tailings,
- Reconstruction of the diversion ditch; and
- Construction of a fence around the property perimeter.

Evaluation of these remedial measures relative to potential site risks is presented in the conceptual site model found in Section 2.2.4 of the RIFS SAP (RMC, 2001).

This SAP is being prepared to complete relevant data gaps identified in the Draft SERA (Syracuse, 2002). The SERA concluded that the Richardson Flat Tailings site is not contributing to increased risks in Silver Creek (Syracuse, 2002). On May 14, 2002 United Park representatives met onsite with EPA and United States Fish & Wildlife Service (USFWS) representatives to discuss the scope of the data collection activities and to determine appropriate locations for the sample collection. It was determined and agreed upon at this site visit that the ecological risk assessment data collection activities would focus on the wetland and pond areas. Furthermore, it was agreed that uncertainties identified in the SERA related to the seeps would not be further quantified. Instead the wetland surface water, sediment, sediment-pore water and biota metal concentrations would be quantified. The SERA over emphasized the potential for the seep water to impact the environment. Very little, if any, of the seep water reaches the wetland area via overland flow.

As part of the focused RI/FS process, United Park has determined that additional remedial measures will be considered as mitigation for any unacceptable risks identified after analysis of the information collected to fill data gaps identified in the draft SERA. Work completed under this SAP will address the remaining data gaps and provide pertinent data for a *focused* assessment of ecological risks. Additional remedial measures that may be considered are:

- Placement of additional clean fill over the impoundment where the existing cover is less than 12 inches. Addition of the fill will allow surface water on the impoundment to be drained to the South Diversion ditch. This will effectively manage areas where tailings may be exposed and may eventually dry up the seeps located at the toe of the embankment.

- Installation of a wedge buttress on the face of the tailings embankment; as part of this work, a drainage system will be installed in the seep area.

- Removal of tailings in certain areas outside of the impoundment and placement of clean fill over other areas of tailings outside of the impoundment.

The above remedial measures will have a profound effect on the release mechanisms, exposure media, exposure route and ecological receptors identified in the Ecological Site Conceptual Model (ESCM) portrayed in Figure 4-1 of the Draft SERA. Specifically the following release mechanisms, exposure media, exposure routes and ecological receptors will be affected by the remedial measures:

- Wind erosion of the tailings for a short time period following active deposition of tailings in the impoundment may have impacted amphibians and reptiles as identified in the ESCM. However, the tailings have been covered for approximately 10 years and additional cover will be placed on tailings in and outside the impoundment. In limited areas east of the impoundment, tailings will be excavated and placed on the impoundment and covered with clean fill. Placement of additional cover soil will significantly reduce or eliminate exposure of amphibians and reptiles to wind blown tailings.

- In areas where the cover thickness over tailings is less than one foot in thickness, additional clean fill will be placed to increase the cover depth to one foot or greater. This will mitigate cap penetration and reduce direct contact and ingestion of soil for birds, mammals, amphibians, reptiles, plants and soil fauna, thereby reducing risks.

- Mixing of the cover soils with tailings will be mitigated by the additional cover soils placed on the entire impoundment. Impacts to sediment from mixing of cover soils with tailings have never likely been a problem at the site. Impacts to sediments in the past were the result of oversteepened side slopes on the diversion ditch channel. The angles of the sideslopes were reduced and revegetated in the early 1990's. Sediment and ecological receptor impacts will be quantified in this SAP.

- Impacts to surface water from rain and snow precipitation and interaction with tailings will be mitigated by source removal activities identified above. Surface water data from the RI indicate that for the most part the Site waters meet applicable state water quality standards.

However, in the upper section of the diversion ditch zinc concentrations increase to levels above those water quality standards. Based on this and other data (depth of cover, tailings thickness, etc) collected during the RI those tailings located east of the diversion ditch near surface water monitoring stations RF-11 and RF-12 will be excavated. Other tailings found outside of the impoundment may either be excavated and/or covered with additional clean fill. However, this release mechanism and subsequent exposure routes and ecological receptors will be evaluated as outlined in this SAP.

- Any impacts to groundwater from leaching within the impoundment will be further mitigated by the additional covering of the impoundment with low permeability clean fill. The cover will be graded to direct surface water to the South Diversion Ditch. The low permeability of the existing and additional cover along with improved surface water runoff design will substantially reduce any infiltration of water into the impoundment. This will further reduce the potential for leaching and could potentially reduce the amount of subsurface water in the impoundment. The reduction in the subsurface water may reduce and possibly eliminate the seeps located in the embankment area.

2.3 Previous Site Investigations

Previous site investigation activities have focused on nature and extent of contamination within and nearby the Study Area (Figure 2.0). United Park has submitted, on December 17, 2002, a Draft Remedial Investigation report summarizing previous data collection activities as well as data collection conducted as part of the AOC.

Previous studies to characterize the wetland area at the site have been limited to sediment, surface and groundwater data collection. In 1992, EPA's contractor, Ecology and Environment (E&E) collected four (4) sediment samples in the wetland area, two (2) surface water samples upgradient and downgradient of the wetland area and two (2) groundwater samples upgradient of the wetland area (E&E, 1993). Sediment sample data from the E&E investigation revealed elevated levels of lead, zinc, and copper.

As part of the RI, surface and groundwater data were collected for a fifteen-month period, beginning in March of 2001. These data indicate that water leaving the diversion ditch meets all applicable water quality standards. Groundwater data from piezometer RT-7 meets all applicable water quality standards, and groundwater data from monitoring well RT-12 exceeds water quality standards for aluminum, cadmium, copper, lead, antimony, and zinc. Groundwater data from monitoring well RT-11 upgradient of the wetland area exceeds water quality standards for antimony, cadmium, lead and zinc. Silver Creek surface water upgradient of the wetlands exceeds water quality standards for zinc and cadmium. Surface water in Silver Creek downgradient of the wetland exceeds water quality standards for only zinc. Surface water from the diversion ditch provides some dilution to the cadmium and zinc concentrations in the Silver Creek surface water downstream of the confluence with the diversion ditch.

The South Diversion Ditch that flows through the pond is a stormwater and groundwater interception ditch that was constructed to divert storm and groundwater from entering the tailings impoundment. The ditch was constructed in the early to mid 1970's by Park City Ventures (PCV), however the ditch was constructed with oversteepend bank slopes in tailings over most of its' length. In 1993 United Park reconstructed the ditch laying back the oversteepend bank slopes and covering the slopes and some tailings in the ditch bottom. Since 1993 surface water quality has improved over much of the diversion ditch. The upper reach of the ditch does contain zinc concentrations in surface water that exceed water quality standards. However, the lower portion of the ditch, from RF-5 to RF-6-2 meets water quality standards (RMC, 2002). The pond is located between these two surface water sample locations.

There have been no studies conducted to date on vegetative types or densities within these areas. Nor have there been any studies conducted on the species composition or population demographics of terrestrial and aquatic biota.

2.4 Environmental Setting

The Study Area is roughly 6,570 feet above mean sea level. The study area is located in the Basin and Range physiographic province, approximately 40 miles northwest of Salt Lake City, Utah.

The Study Area is characterized by a cool, dry, semi-arid climate. Long-term meteorological observations have not been kept at the Site. The two nearest meteorological data stations are located in Park City, Utah which is located 500 feet higher in elevation three miles to the southeast in the Wasatch Mountains, and Kamas, Utah located at a similar elevation to the Site and nine miles to the east. The annual precipitation for the Site likely falls in-between the values for the two sites. Annual precipitation at Park City is 21.44 inches of water with an annual average high temperature of 56.3 degrees and an annual average low temperature of 30.8 degrees. Annual precipitation at Kamas is 17.27 inches of water per year with an average annual low temperature of 29.0 degrees and an average annual high temperature of 58.7 degrees (www.wrc.dri.edu, 2001).

Long-term wind data have not been kept in the vicinity of the Site. The prevailing wind direction is from the northwest to southeast as determined by the EPA contractor Ecology and Environment during an air monitoring assessment conducted in 1986 (E&E, 1987).

More comprehensive descriptions of the environmental settings and lists of references are available in the Focused Remedial Investigation Workplan, Sampling and Analysis Plan (RMC, 2000 and 2001, respectively) and the Draft SERA (Syracuse, 2002).

PART I: FIELD SAMPLING PLAN

3.0 SAMPLING PROGRAM, RATIONALE, AND LOCATIONS

The Field Sampling Plan (FSP) for this investigation has been developed to provide rationale and procedures for the collection of samples to assess levels of metals contamination at specified locations in conjunction with plant and invertebrate community indices, bioaccumulation studies, and laboratory sediment toxicity tests.

3.1 Experimental Design and Sampling Rationale

The general objective of this sampling effort is to collect data needed for the evaluation of hazards posed to terrestrial and aquatic receptors by metals in surface water, sediment and sediment pore water, and biota in the pond and wetland. Results from this sampling effort may be used to further evaluate the fate and transport of metals in surface water and in the sediments found in the wetland and pond areas. Fate and transport anaylses have been performed for the site contaminants as presented in the Focused RI submitted to EPA in December 2002.

To optimize predictive capabilities of effects on potential receptors (e.g., development of PRGs) data will be collected in a manner to quantify bioavailable metal concentrations in sediments (i.e., porewater) and in surface water along with those factors that influence bioavailability (e.g., pH, nutrients, and organic matter). The concentration and form of metals in aquatic systems control metal availability and toxicity. The most toxic form of metals is generally the dissolved free metal ion and the least toxic form is associated with particles, which is the reason concentrations in filtered water samples are compared to ambient water quality criteria to assess risk. The dissolved fraction of the metals, such as found in sediment porewater, can be further broken down into species such as free metal ions, sorbed to colloidal material, and complexed with both organic and inorganic ligands. Data collected on the chemistry of filtered surface water and porewater (e.g., metal concentration, matrix ion concentrations, pH, and DOC) can be further used in a chemical equilibrium model such as EPA's MINTEQ, to calculate the concentrations of metal species, and in particular the free metal ion. The calculated concentrations can then be used to identify possible causes of observed toxicity.

Measures of exposure will also include plant, invertebrate, and fish tissue concentrations of metals. These tissue data will be used in food chain models to assess exposure of and risk to wildlife species dependent on food resources in the wetland and pond. Direct effect measures will include invertebrate toxicity testing of sediments, benthic macroinvertebrate community analysis, and wetland plant community analysis.

3.2 Sample Media and Parameters

All sampling described below is required to achieve the project objectives. The focus of sample collection activities proposed in this SAP is evaluation of the following environmental media in the wetland area west of the main embankment, in the ponded area at the terminus of the diversion ditch, and in corresponding habitats at reference sites. Table 3.0 summarizes the sample media, parameters to be measured, and how these data will be used in the risk characterization.

Table 3.0. Sampling Objectives and Risk Characterization Approach

Media/Parameters	Sampling objectives	Risk Characterization Approach
Surface water: Dissolved metals and water quality parameters	- Determine exposure of aquatic invertebrates, fish, and wildlife receptors. - Determine potential bioavailability of metals.	- Compare dissolved metals concentrations with Utah state water quality standards to estimate risk to water column invertebrates and fish. - Compare exposure of wildlife from ingestion of contaminated drinking water to toxicity reference values.
Sediment: Total metals and compositional properties	- Determine exposure of benthic macroinvertebrates, fish, wildlife, and wetland plants. - Determine the contribution of metals in sediments, if any, to the metal loading in surface water.	- Interpret sediment chemistry data relative to toxicity test and macroinvertebrate data in the Triad assessment. - Compare exposure of wildlife from ingestion of contaminated sediment and food to toxicity reference values.
Pore water (sediment): Metals and water quality	- Determine exposure of benthic macroinvertebrates. - Determine potential bioavailability of metals. - Determine if the sediments are a source or sink for metals in the surface water.	- Interpret pore water chemistry data relative to toxicity test data and macroinvertebrate data in the Triad assessment. - Use data in a chemical equilibrium model (e.g., MINTEQ) to calculate the concentrations of metal species and free metal ion
Sediment: Toxicity test responses	- Evaluate exposure-response relationships for aquatic macroinvertebrates. - Estimate risk to aquatic macroinvertebrates.	- Determine statistically significant toxicity relative to reference area sediments (and > 25 % response) and interpret as part of Triad assessment.

Table 3.0. Sampling Objectives and Risk Characterization Approach (cont.)

Media/Parameters	Sampling objectives	Risk Characterization Approach
Sediment: Benthic macroinvertebrate community indices	- Evaluate exposure-response relationships for aquatic macroinvertebrates. - Estimate risk to aquatic macroinvertebrates.	- Determine statistically significant differences in macroinvertebrate indices between site stations and reference area stations and interpret as part of Triad assessment.
Sediment and surface water: Wetland plant community indices	- Evaluate exposure-response relationships for plants. - Estimate risk to plants.	- Determine statistically significant differences in plant community indices between site stations and reference area stations and interpret compositional differences relative to literature.
Tissue: Vegetation, invertebrate, and fish	- Determine exposure of wildlife receptors. - Evaluate uptake of metals from the sediment, surface water and pore water. - Determine bioaccumulation of metals.	- Compare exposure of wildlife from ingestion of contaminated sediment and food to toxicity reference values.

Section 4.5 (and Table 4.1) discusses in detail the field sampling and data collection. Metals data will be used to estimate exposure of ecological receptors, while biological data are used as measures of response.

The metals to be analyzed will be a subset of the EPA Target Analyte List (TAL) parameters. Although EPA, in the Draft SERA, recommended that most media at the Site be analyzed for TAL parameters, United Park has determined data are available for all of the TAL metals. Specifically, those data can be found in the 1985 E&E report, "Analytical Results Report, Richardson Flat Tailings", TDD R8-8508-7 and in the Draft RI report, dated December 17, 2002. EPA should consider use of these data to address some of the obscure analytes such as cobalt, thallium and vanadium.

Although some of the metals on the TAL were not analyzed during the Focused RI/FS, X-Ray Diffraction (XRD) data for the Richardson Flat tailings indicate it would be unlikely that metals such as barium, beryllium, boron, cobalt, nickel, thallium and vanadium would be present at above-background concentrations. The XRD results, summarized in Table 5-2 of the draft Focused RI Report, do not indicate the presence of minerals containing any of these metals. In fact, the bulk of the tailings mineralogy consists of quartz (SiO_2), calcite ($CaCO_3$) and dolomite ($CaMg(CO_3)_2$), which provide little opportunity for elemental substitution of these particular TAL metals in their structures. Although limited substitution could occur in these minerals (e.g., barium or cobalt for calcium in calcite), the resulting metals concentrations in the tailings would be low. Typically these substitutions represent only minor impurities.

Some substitution could also occur in some of the less prevalent minerals found in the tailings. For example, pyrite (FeS_2) with small amounts of nickel and cobalt substituting for iron is not uncommon (Deer, Howie and Zussman, 1966). In the case of galena (PbS), substitution of other atoms for lead is not very extensive but may include small amounts of metals including antimony, arsenic, cadmium, silver, thallium and zinc (Deer, Howie and Zussman, 1966). Other elements, such as beryllium (beryl), boron (borates), and vanadium (vanadates) are more typically found in very particular minerals not found in these tailings.

Therefore, the metals targeted for this investigation are: aluminum, antimony, arsenic, cadmium, chromium, copper, iron, lead, manganese, mercury, selenium, silver, and zinc. Based on the mineralogy and geochemistry of the ore deposits in the Park City area (or similar Lead-Zinc-Silver ore deposits studied elsewhere), high concentrations of additional metals are not expected.

3.3 Sampling Locations

Figure 3.0 shows the locations of the sampling stations at the site. Sampling locations were determined during the site visit with EPA and USFWS representatives on May 14, 2002. Eight sampling sites were selected in the wetland area and two sampling sites were selected in the pond.

Six sampling sites were selected in the wetland channel prior to the convergence with the Silver Creek water and two sampling sites were selected after the convergence with Silver Creek. Data from the two sample sites in the area where Silver Creek converges with the wetland flow will be used to evaluate possible ecological impacts in the wetland. These data will not be used to evaluate impacts to Silver Creek. Impacts to Silver Creek are being evaluated under the Upper Silver Creek Watershed Stakeholder Group. Data from the six sample sites located in the wetland, prior to the convergence with Silver Creek, will be used to evaluate impacts to the wetland from the Richardson Flat Tailings impoundment.

Two sampling locations were selected in the pond area as shown on Figure 3.0. The pond is relatively small with an area of one (1) acre. The pond sampling locations were selected to represent an average of the pond surface water and sediment characteristics.

In addition to the on-site stations, two stations will be sampled in reference areas located in mineralized areas without mining activities: one in a wetland habitat with physical properties approximately similar to the on-site wetland, and one in a pond habitat. Selection of appropriate reference sites will be conducted as part of the habitat characterization of the site (refer to Section

4.2) and with concurrence from EPA and USFWS representatives. One sample location will be located at each reference site.

Sampling stations for all elements of this investigation (i.e., the surface water assessment, the sediment Triad, plant community analysis, and tissue sampling) will be collocated at the sites shown in Figure 3.0 and described above. Discrete samples will be collected for metals analysis in surface water, porewater, and sediment, and sediment toxicity testing and macroinvertebrate community analysis. Grid sampling for the plant community analysis will be centered on points collocated with the media samples. Tissue samples for some biota (i.e., macroinvertebrates and fishes) will be composited across stations to represent an average for wetland and an average for pond habitats at both the site and the reference area. This approach will be used to mimic the exposure regime of wildlife receptors, which range across areas at least as big as the wetland and the pond.

4.0 FIELD ACTIVITY METHODS AND PROCEDURES

The following field activities and procedures will be employed for this project (see Section 5.7 for laboratory analytical methods):

- Site Mobilization
- Surveying and Habitat Characterization
- Mobilization of Equipment, Supplies, and Containers
- Equipment Decontamination
- Field Sample Collection
  - — Surface Water Sampling
  - — Sediment Sampling
    - ⇒ Sediment Samples for Physical-Chemical Analysis
    - ⇒ Sediment Toxicity Testing Sampling
    - ⇒ Benthic Macroinvertebrate Community Survey
    - ⇒ Benthic Macroinvertebrate Tissue Sampling for Metals Analysis
  - — Pore Water Sampling
  - — Plant Tissue Sampling for Metals Analysis
  - — Fish Tissue Sampling for Metals Analysis; and
  - — Plant Community Survey
- Investigation-Derived Waste.

Referenced SOPs are included in Appendix A.

4.1 Site Mobilization

RMC will identify and provide all necessary personnel, equipment and materials for mobilization and demobilization to and from the site for the purpose of conducting the sampling events. Equipment mobilization also entails ordering and purchasing of equipment and supplies. A complete inventory of available equipment and supplies will be conducted prior to initiating the field activities and any additional required equipment or supplies will be obtained.

4.2 Surveying and Habitat Characterization

Global positioning system (GPS) receivers will be used to locate all sampling locations in the field.

A habitat characterization will be conducted concurrent with sampling activities. This will include a physical characterization of the site and a characterization of the water quality. The physical characterization will include documentation of land use, a description Silver Creek, the diversion ditch, and pond; and documentation of the riparian and wetland vegetation. Waterbody parameters such as width, depth, flow, and substrate will also be recorded. The water quality characterization will include field and laboratory measurements of temperature, pH, dissolved oxygen, and turbidity (refer to Table 4.1). The information collected during the habitat characterization will provide information regarding the ability of the waterbodies and wetland to support a healthy ecological community, and regarding the presence of stressors to the ecosystem.

A vegetation covertype map will be developed as part of the habitat characterization for the site and surrounding vicinity. Qualitative observations of wildlife usage of the site will also be recorded. This information will be used in the development of the conceptual site model (i.e., identification of exposure pathways and ecological receptors) for the ecological risk assessment.

As part of the habitat characterization, reference sites will be selected. The reference areas will consist of one wetland habitat and one pond habitat. Reference areas will be selected that are in non-impacted, mineralized areas that have similar physical conditions to the onsite wetland and pond, such as soil type, hydrology, topography, and vegetative community.

4.3 Equipment, Supplies and Containers

Equipment and supplies necessary to support the field activities are summarized in Table 4.0. This table separates field items into the following categories: sampling, health and safety, equipment and personal decontamination, and general field operations.

Sample containers and any required preservatives will be supplied by the laboratories or purchased from approved vendors. All sample containers will be pre-cleaned and traceable to the facility that performed the cleaning. Sample containers will not be cleaned in the field. Surface and pore water containers will be triple rinsed in the field with sample media prior to filling.

4.4 Equipment Decontamination

All sampling equipment will be decontaminated prior to use at each station and between media types. Equipment decontamination procedures outlined in *Sampling Equipment Decontamination* (RMC SOP 6 provided in Appendix A) will be used in this sampling program. Equipment decontamination will be performed by placing the sampling equipment in a bucket filled with deionized (DI) water and non-phosphate soap, and removing any visible residual material from the sampling equipment with a brush. Any residual soap or debris will be removed by pouring DI water over the equipment. Sampling equipment will then be double rinsed with deionized water. Upon completion of this procedure, all equipment will be air dried and stored in a "clean" vessel or wrapped with foil until ready for use. Disposable, one use, sampling equipment will be used to the extent possible.

4.5 Field Sampling and Data Collection

Table 4.1 provides a summary of the number of samples and analyses that will be collected during the field investigation. The sample volumes, containers, and preservation requirements for these samples are specified in the QAPP (Part II). Samples for chemical analysis will be identified as follows: surface water samples will be designated with SW identifier, sediment with a SD, sediment pore water with a PW, vegetation with a VEG identifier, benthic macroinvertebrates with a BMI identifier, and fish with a FI identifier. Sediment bioassay samples will be identified with SD-BIO, benthic macroinvertebrate samples for community analysis with a BMI-COM, and vegetation community stations with a VEG-COM identifier. The methods that will be used to collect the samples are discussed below.

4.5.1. Surface Water Grab Samples

Surface water samples will be collected at a minimum of eight (8) locations in the wetland area and two (2) locations in the pond area as depicted on Figure 3.0, and at two (2) reference stations in corresponding habitats. The on-site sampling locations were selected during the May 2002 site visit by EPA and USFWS representatives and also based on data uncertainties identified in the Draft SERA.

The samples will be collected according to the SOP, *Surface Water Sampling* (RMC SOP 1), presented in Appendix A. Field analytical parameters and procedures are shown on Table 4.1 of this SAP. Surface water samples for dissolved metals analyses will be filtered in the field prior to sample preservation (RMC SOP 1, Appendix A). Sampling locations will be logged with a Global Positioning Survey (GPS) unit.

4.5.2. Sediment and Benthic Macroinvertebrate Samples

Surface sediment and macroinvertebrate samples for analysis of sediment physical-chemical parameters, toxicity, benthic macroinvertebrate tissue chemistry, and benthic macroinvertebrate community structure will be collected at two (2) locations in the pond, at eight (8) locations in the wetland, and at two reference site locations. All sampling locations will be co-located with the surface water sampling locations (Figure 3.0).

An Ekman grab sampler will be used to collect sediment material from the 0–10 cm horizon at each station for chemical analyses, toxicity testing, and macroinvertebrate tissue analyses. For collection of macroinvertebrates for community analysis, the contents of entire grab samples will be retained. If plant roots or other material in the sediments precludes use of a grab sampler, sediments will be collected by using a drive rod check-valve corer or other similar coring equipment. The 0–10 cm depth horizon designated for each analysis would then be exuded from each core. For collection of benthic community samples, the sampling device should be consistent among stations, so a reconnaissance of stations will be needed to assess which device is most appropriate. Procedures for sampling are described in the SOPs entitled *Surface Sediment Sampling Using an Ekman Grab Sampler* (Exponent SOP SD-05); and *Sediment Coring Using a Drive Rod Check-Valve Corer* (Exponent SOP SD-10) provided in Appendix A.

Each sediment sample will be evaluated for acceptability based on the following criteria:

- Overlying water is present and not excessively turbid
- The sediment surface is relatively undisturbed
- The planned penetration depth of the grab sampler or push corer is achieved.

If a sample fails to meet any of the acceptance criteria, it will be rejected and discarded away from the station. Multiple cores or surface grabs may need to be collected from each station to provide sufficient material for all specified analyses.

4.5.2.1. Sediment Chemistry and Toxicity Testing Samples

Sediment samples to be analyzed for physical-chemical parameters and for toxicity will be splits of a field homogenate at each station. The 0-10 cm depth horizons collected from each sediment sample for sediment chemistry and toxicity testing will be transferred into a stainless-steel bowl. The sample(s) from a given station will be mixed with large stainless-steel spoons to achieve a uniform texture and color. The homogenized sample will be subsampled and transferred to the appropriate sample containers (Table 4.1). Samples designated for acid-volatile sulfide analyses will be collected prior to homogenizing the sample. Large artifacts such as rocks and twigs will be removed from the sample during homogenization. The relative amount and types of material removed will be noted in the field logbook.

The samples will be placed in glass or polyethylene containers and kept in coolers on ice (4 degrees Celsius) until transfer to a refrigerator at the laboratory. The samples will be stored in glass or polyethylene containers and kept at 4 degrees Celsius. All samples will be analyzed as bulk samples.

4.5.2.2. Benthic Macroinvertebrate Samples

The benthic macroinvertebrate community assessment and tissue analyses investigation consists of the following tasks:

- **Macroinvertebrate Community Analysis**—Grab sampling will be performed to evaluate benthic community composition, including taxa abundance and distribution. Ten (10) on-site sampling stations and two (2) reference stations will be co-located with the other sediment and surface water sampling stations. Care will be taken to collect benthic macroinvertebrate samples from locations that were not disturbed by previous sample collection activities.

- **Macroinvertebrate Tissue Sampling**—Sampling and analyses of metals concentrations and percent moisture in dominant macroinvertebrate taxa (e.g., amphipods and chironomids) will be completed to evaluate metals bioaccumulation and potential for transfer to higher trophic levels. Ten (10) on-site sampling stations and two (2) reference station will be co-located with the other sediment and surface water sampling stations. Tissue samples for macroinvertebrates will be composited across stations to represent an average for wetland and an average for pond habitats at both the site and the reference area. This approach will be used to mimic the exposure regime of wildlife receptors, which range across areas at least as big as the wetland and the pond.

Sampling to characterize benthic community composition will be completed using an Ekman grab sampler (preferred) or a coring device. Each grab sample will be evaluated for acceptability based on the criteria listed above. Five replicate grab samples of surface sediments (0-10 cm) will be collected from each of the 12 sampling locations. If it is necessary to use a coring device instead of a grab sampler (in order to cut through plant material in the sediment), 15 replicate cores (0–10 cm depth horizon) will be collected at each station. Whichever sampling device is chosen for the investigation, the same device should be used consistently among all stations.

Each replicate sample, consisting of sediment and the overlying water, will be strained through a sieve (600 µm) to isolate benthic organisms. Retained material from each grab sample will be transferred to an appropriate sample container and preserved in the field for later sorting and identification in a taxonomic laboratory. Detailed sampling procedures for the collection of grab samples and processing of macroinvertebrate samples are presented in the SOPs, *Surface Sediment Sampling Using an Ekman Grab Sampler*; and *Sediment Coring Using a Drive Rod Check-Valve Corer* (Exponent SOPs SD-05 and SD-10) (Appendix A).

Sampling to collect macroinvertebrate tissues for metals analyses will be completed using a grab sampler (Ekman) or corer to collect surface sediments (approximately 0–10 cm depth) from 10 on-site locations and 2 reference locations. The number of samples required to collect tissues for constituent analyses will vary at each location according to the abundance of amphipods and chironomids in the surface sediment. Sediment collection at each location will continue until the minimum mass requirement for tissue analyses (30-50 g organisms) is obtained at each location, or until a reasonable effort has been expended to obtain the sample. Sediments and overlying water from each grab sample will be strained through an appropriately sized screen (600 µm) with gentle streams of water. The contents of each sieve will be combined into a large collection container, covered with water, and sorted at the field laboratory to isolate the target macroinvertebrate sample groups. Target organisms will be collected and preserved on ice for subsequent processing and transfer to the analytical laboratory. Detailed sampling procedures for collecting and processing macroinvertebrate tissues and sediment samples are included in the SOPs, *Benthic Macroinvertebrate Sampling using a Grab Sampler (*Exponent SOP BI-12) and, *Aquatic Invertebrate Processing Procedures* (Exponent SOP BI-11) (Appendix A).

4.5.3. Sediment Porewater

Sediment porewater will be collected using a micro push point at each of the ten (10) sampling locations on-site and from two (2) reference site locations (Figure 3.0). A porewater sample will be collected assuming there is a positive flux of groundwater to the wetland or pond at each station. Porewater samples will be collected following methods in the porewater sampling SOP, *Porewater Sampling from a Micro Push Point or Mini Piezometer* (SOP #SRC-OGDEN-01) (Appendix A). Sediment porewater samples for dissolved metals analyses will be filtered in the field prior to sample preservation (RMC SOP 1, Appendix A). Some modifications to the porewater sampling method may be required based on the sediment particle size and other sediment characteristics encountered in the field at the time of sampling. Any deviations from the SOP that may be required to ensure sample representativeness will be documented in the field logbook.

4.5.4. Plant Tissue Samples for Metals Analysis

Plant tissue samples will be collected from the ten (10) sampling locations on-site and from two (2) reference site locations (Figure 3.0). Sampling methods will follow Exponent SOP BI-13, *Vegetation Sampling* and Exponent SOP BI-01, *Decontamination of Equipment—Tissue* (Appendix A). Methods not addressed in these SOPs will follow EPA/ERT SOP #2037, *Terrestrial Plant Community Sampling* and Exponent BI-13, Vegetation Sampling (Appendix A).

A single dominant plant species that serves as a food source for terrestrial receptors will be targeted for tissue sampling. The dominant forage plant species will be determined by conducting a qualitative survey of the plant species in the on-site wetland and pond. A qualified botanist will record visual cover estimates at each sampling location. The forage species with the highest average cover within each plant community will be selected for plant tissue collection. In the pond, plant tissue samples will be collected from plants nearest the bank assuming an emergent species is the plant species selected for analysis. If the species selected for analysis is submerged aquatic vegetation or if emergent species are not present in the pond, then a submerged vegetation species will be sampled to represent the plant species for the pond.

A 1 m^2 PVC tube quadrant frame will be used to delimit each of the individual sampling points. Tissue from several individual plants of the dominant herbaceous plant species may have to be collected at each location to obtain enough sample volume. Vegetation sampling locations will be co-located with the surface water and sediment sampling sites. Herbaceous plant tissue sampling will involve the collection of the aboveground biomass only, utilizing a pair of stainless steel scissors.

The wetland plant community likely features sedges, rushes, willows, and various other woody scrub/shrub species. For this community type, a larger 10 m^2 sampling plot will be utilized (2.5 meters by 4 meters). If these plots are dominated (in terms of percent cover) by woody shrubs (such as Rocky Mtn. willow), then branches will be cut from the dominant shrub species using pruning shears. The cut branches will then be stripped of leaves, buds, and fruiting structures (if present) for the tissue sample. Several plants within the large plots will be sampled in order to provide a representative mass of plant tissue from the dominant species. If the plot is dominated by herbaceous vegetation, such as sedges, then the dominant species will be collected in the same manner as herbaceous matter was collected as described above (i.e., by cutting aboveground shoot mass with stainless steel scissors).

One-gallon, resealable plastic bags will be used to contain samples of the dominant vegetation from each location. The samples will be placed on ice in coolers, transported to the laboratory and transferred to a refrigerator at 4 degrees Celsius until analysis.

4.5.5. Fish Tissue Samples for Metals Analysis

Composite samples of the two most abundant species of forage fishes will be collected from the wetland and pond on-site (Figure 3.0) and from two stations at the reference sites. Fish tissue samples will be composited across stations to represent an average for wetland and an average for pond habitats at both the site and the reference area. This approach will be used to mimic the exposure regime of wildlife receptors, which ranges across areas at least as big as the wetland and the pond.

The species targeted for sampling will be those that may potentially serve as a food resource for piscivorous birds and mammals. Because data are not available on the fish community composition at the site and reference areas, a reconnaissance survey will be conducted to provide data for selection of target species for the investigation.

The wetland, the pond, and the reference sites will each be considered a "station area" for fish sampling because fish move among specific locations designated for other sample analyses. At each of the four station areas (i.e., site wetland, site pond, reference wetland, and reference pond), 15 individuals of each of the two dominant fish species will be collected for analyses. Only fish from 2-4 inches in total length will be retained for analyses in order to target the size classes available to a wide range of predators and to limit variability of the data due to any age/size-related factors. Species will be maintained separately for analysis. Three composite samples of five individual fish of each species will be formed at each station. The individuals comprising each composite sample will be selected so that the average total length of fish does not differ significantly between replicate composite samples (by species).

Depending on the physical characteristics of the sampling locations, fish will be collected using electrofishing units, beach seines, minnow traps, or a combination of techniques. Procedures for operation of each type of equipment are summarized below. Detailed procedures are provided in NYSDEC (1999a) and in *Fish Collection Procedures Using an Electroshocker* (Exponent SOP BI-04) (Appendix A).

The electrofishing unit sends an electric current through the water, temporarily stunning the fish. The stunned fish are then collected with a scap net. Because the electrofishing unit generates electric current, several precautions must be taken to avoid electrocution during sampling. Electrofishing will only be conducted by technicians who are familiar with the appropriate safety procedures, and all equipment will be maintained and operated according to the manufacturer's instructions. All persons in the sampling crew must wear hip boots or chest waders as a safety precaution.

Beach seines are manually dragged along the shore to collect fish in shallow waters. Minnow traps are passive collection devices (i.e., fish enter the traps but cannot escape) that must be anchored in place and set for several hours.

The following information will be recorded as soon as possible after sample collection for all fish collected:

- Weight and total length measurements
- Reproductive state
- Presence of grossly visible abnormalities.

Procedures for determining length and weight of fish are described in NYSDEC (1999a) and in *Fish Processing Procedures* (Exponent SOP BI-08) (Appendix A).

After length and weight measurements have been made, fish will be double-bagged in two plastic Ziploc® bags containing a sample identification label. Fish for composite samples will be bagged together in two plastic Ziploc® bags to represent one sample for analytical purposes.

At the field office, samples will be packaged on ice in coolers and shipped by local courier or overnight delivery service to the analytical laboratory for chemical analysis. The analytical laboratory performing chemical analyses on whole-body samples will be responsible for sample homogenization and (if appropriate) transferring sample aliquots required for chemical analysis to the appropriate laboratories.

4.6 Plant Community Metrics

Plant community observations will be made in accordance with EPA/ERT SOP #2037, *Terrestrial Plant Community Sampling* (Appendix A). A qualitative assessment of the type and extent of the wetland habitat will be conducted by a qualified botanist. Prior to collecting sediment and plant tissue samples from the plots established in upland and riparian areas in Phase II, a botanist will record data based on visual observations within each plot. These data will include a record of signs of vegetative stress (wilting, browning, stunted growth, chlorosis, grazing evidence, etc.), and habitat characteristics (spatial arrangement of species, foliage density, light penetration characteristics, moisture availability, slope aspect, sediment characteristics, etc.).

A quantitative survey of the wetland plant community will be conducted at the 12 co-located sediment and surface water stations (Figure 3.0). The botanist will collect data that will yield measures of plant diversity and community similarity, including: species composition, species-specific cover and frequency, species richness, and species evenness in the sample plots at each station. A 1 m^2 PVC tube quadrat frame will be used to delimit each of the individual sampling plots at each station. Three replicate samples will be taken at each station for species counts. The quadrat will be randomly placed by an individual standing at the station and throwing the quadrat over his/her shoulder. Species composition and total species richness measurements will also be made at each station by identifying species within a 10 m^2 sampling plot centered on the station.

4.7 Investigation-Derived Waste

Investigation-derived waste (IDW) generated during this study will be handled in accordance with OERR Directive 9345.3-02 *Management of Investigation-Derived Wastes During Site Inspections* (EPA, 1991). Collecting only the volume of material needed to satisfy laboratory analytical requirements will minimize the generation of IDW. Any excess material will be discarded at the sample collection point.

PART II: QUALITY ASSURANCE PROJECT PLAN

5.0 PROJECT MANAGEMENT

The QAPP (Part II of this SAP) for the Richardson Flat ecological risk assessment has been developed in accordance with EPA QA/R-5 guidance for preparing QAPPs (EPA, 1997). This section covers the basic area of project management, including the project organization, background and purpose, project description, quality objectives and criteria, special training, and documentation and records.

5.1 Project Organization

Organization and responsibilities specific to this investigation are discussed in this section. Laboratory services will be provided by an EPA approved laboratory, which will analyze the surface water, sediment, sediment-pore water, vegetation and biological tissue samples for metals.

For this data collection effort, key management personnel are as follows:

Individual	Role/Responsibility
Kerry Gee	United Park Project Manager
Jim Fricke	RMC Site Manager
Linda Ziccardi	Exponent Ecotoxicologist
Jim Christiansen	EPA Remedial Project Manager
Dale Hoff	EPA Regional Ecotoxicologist
Dan Wall	EPA/USFWS Liaison
Mohammad Slam	UDERR
Gary Colgan	CH2M Hill – QA Official

The management team consists of United Park personnel with assistance from RMC and other environmental consulting firms as needed. Figure 5.0 shows the chain-of-command for the project managers, engineers, and quality assurance officials responsible for managing the Richardson Flat Tailings Site Ecological Risk Assessment SAP.

United Park's environmental Project Manager for the Site is Kerry Gee, who will be responsible for all project management and communication with the regulatory agencies. Jim Fricke of RMC, Salt Lake City, Utah, leads United Park's environmental project consultant team and will be the Site Manager, who will be responsible for implementation of the SAP. Todd Leeds, of RMC, is the Field Manager who will be responsible for all field activities related to this document. Wesley McDonald, RMC, is the Site Safety Officer, who will be responsible for visitor sign in and ensure that all site visitors comply with the HASP.

The EPA Project Coordinator is Jim Christiansen, Region VIII, Denver, Colorado. The Utah

Department of Environmental Remediation and Response (UDERR) Project Manager is Muhammad Slam. The EPA Project Coordinator and the UDERR Project Manager work cooperatively to oversee the work being performed at the Richardson Flat site.

Mr. Gee, as Project Manager, is responsible for the overall management and coordination of the following activities:

- Coordination with EPA/UDERR regarding the status of the project;
- Providing oversight of the subcontractors;
- Reviewing monthly status reports;
- Supervising production and review of deliverables;
- Tracking work progress against planned budgets and schedules;
- Informing EPA/UDERR of changes in the Workplan, SAP, HASP and/or other project documents;
- Notifying EPA/UDERR immediately of significant problems affecting the quality of data or the ability to meet project objectives;
- Procuring subcontractors to provide sampling and analytical support;
- Providing oversight of report preparation;
- Organizing and conducting a field planning meeting.

Mr. Fricke, as the Site Manager, is responsible for the following:

- Preparing monthly status reports;
- Coordinating with the laboratory regarding the analytical, data validation, and Quality Assurance/Quality Control (QA/QC) issues related to sample analysis;
- Reviewing analytical results and deliverables from subcontractors;
- Incorporating changes in the Workplan, SAP, HASP, and/or other project documents;
- Scheduling personnel and material resources;
- Implementing field aspects of the investigation, including this SAP and other project documents;
- Implementing the QC measures specified in the QAPP in this and other project documents;
- Implementing corrective actions resulting from staff observations, QA/QC surveillance, and /or QA audits;
- Providing oversight of data management;
- Coordinating and overseeing the efforts of the subcontractors providing sampling and analytical support;
- Scheduling and conducting field work;
- Notifying the subcontract analytical laboratory of scheduled sample shipments and coordinating work activities;
- Gathering sampling equipment and field logbooks, and confirming required sample containers and preservatives.
- Maintaining proper chain-of-custody forms and shipping of samples to the analytical laboratory during sampling events;

- Ensuring that sampling is conducted in accordance with procedures detailed in this SAP and that the quantity and location of all samples meet the requirements of the SAP; and
- Identifying problems at the field team level; resolving difficulties in consultation with the QA/QC staff; implementing and documenting corrective action procedures at the field team level; and providing communication between the field team and United Park management.

The roles and responsibilities of other field team members will be to assist the Site Manager with sampling activities, sample handling, and overall documentation. Oversight activities including sampling to be conducted by EPA's on-site contractor will be coordinated between the EPA Project Coordinator and United Park's Project Manager. EPA's on-site contractor and the Site or Field manager will work together to coordinate sampling efforts.

5.2 Quality Assurance/Quality Control Organization

The Quality Assurance Official (QAO) is Gary Colgan, with CH2M Hill, who is responsible for the quality assurance/quality control of the data that are generated during implementation of the SAP. Mr. Colgan will report any QA/QC problems to the Site Manager. As the QAO, he will be responsible for the following:

- Reviewing and approving project specific plans;
- Directing the overall project QA/QC program;
- Maintaining QA/QC oversight of the project;
- Reviewing QA/QC sections in project reports, as applicable;
- Reviewing QA/QC procedures applicable to this SAP;
- Auditing selected activities of this project performed by RMC and subcontractors, as necessary;
- Initiating, reviewing, and following up on response actions to address QA/QC problems, as necessary;
- Consulting with the Site Manager and/or Project Manager, as needed, on appropriate QA/QC measures and corrective actions;
- Arranging performance audits of measurement activities, as necessary; and
- Providing written reports on QA/QC activity to the Project Manager and Site Manager.

5.3 Background and Purpose

Site background information for the Richardson Flat Site is provided in Section 2.0 of this SAP. The purpose and objectives of the work assignment are discussed in Section 1.1 of this SAP. The purpose of this QAPP is to provide guidance to ensure that all environmentally related data collection procedures and measurements are scientifically sound and of known, acceptable, and documented quality conducted in accordance with the requirements of the project.

5.4 Project Description

The QAPP addresses field work, data collection and laboratory analyses performed for this work assignment. Detailed project descriptions are outlined in the FSP sections above.

5.5 Data Quality Objectives (DQOs) and Criteria for Measurement

This section provides internal means for control and review so that environmentally-related measurements and data collected in this study are of known quality. The subsections below describe the DQOs (Section 5.5.1) and data measurement objectives (Section 5.5.2).

5.5.1. Data Quality Objectives

The DQO process is a series of planning steps based on the scientific method that are designed to ensure that the type, quantity, and quality of environmental data used in decision-making are appropriate for the intended purpose. The EPA has issued guidelines to help data users develop site-specific DQOs (EPA, 1994b). The DQO process is intended to:

- Clarify the study objective;
- Define the most appropriate type of data to collect;
- Determine the most appropriate conditions from which to collect the data; and
- Specify acceptable levels of decision errors that will be used as the basis for establishing the quantity and quality of data needed to support the design.

The goal of the DQO process is to help assure that data of sufficient quality are obtained to support remedial response decisions, reduce overall costs of data sampling and analysis activities, and accelerate project planning and implementation. Data Quality Objectives are summarized in Table 5.0.

The DQO process specifies project decisions, the data quality required to support those decisions, specific data types needed, data collection requirements, and analytical techniques necessary to generate the specified data quality. The process also ensures that the resources required to generate the data are justified. The DQO process consists of seven steps, of which the output from each step influences the choices that will be made later in the process. These steps include:

Step 1: State the problem;
Step 2: Identify the decision;
Step 3: Identify the inputs to the decision;
Step 4: Define the study boundaries;
Step 5: Develop a decision rule;
Step 6: Specify tolerable limits on decision errors; and
Step 7: Optimize the design.

During the first six steps of the process, the planning team develops decision performance criteria (DQOs) that will be used to develop the data collection design. The final step of the process involves developing the data collection design based on the DQOs. A brief discussion of these steps and their application to this project is provided below.

Step 1: State the Problem

The purpose of this step is to describe the problem to be studied so that the focus of the study will be unambiguous. The wetland and pond areas currently have no remedial action planned to directly reduce potential exposure of metals to riparian receptors. The Draft SERA utilized conservative risk estimates because there was no site specific information available to determine the nature and extent, sediment based toxicity measures of bioavailability and toxicity to determine if more remedial activity is necessary; and if it is necessary, what clean-up level should be established.

Step 2: Identify the Decision

This step identifies what questions the study will attempt to resolve and what actions may result. The principal study question for these areas is: "Are adverse effects observable in terrestrial and aquatic receptors at this site?" The question to be answered using the data is: "Are the concentrations of metals in the sediments correlated with uptake metals in vegetation and aquatic species, and do these levels correlate with flora and fauna demographics and toxicity on the site?"

Step 3: Identify the Inputs to the Decision

The purpose of this step is to identify the information that needs to be obtained and the measurements that need to be taken to resolve the decision statement. Based on the study questions, the following information is required:

- The concentration of dissolved metals in the surface water;
- The concentration of metals in the sediments;
- The bioavailable concentration of metals in the sediments (i.e., porewater);
- The concentration of metals in the wetland vegetation;
- The concentration of metals in benthic macroinvertebrate tissue;
- The concentration of metals in fish tissue;
- The toxicity of bioavailable metals in the wetland and pond sediments to invertebrates;
- Plant and benthic community indices in the wetland and pond areas.

Step 4: Define the Boundaries of the Study

This step defines the spatial boundaries of the study. The entire project will be performed within the wetland and pond area as shown on Figure 3.0.

Step 5: Develop a Decision Rule

The sampling phase decision process consists of the following steps:

1) Assess whether the data are usable based on the data validation and evaluation processes. If yes, continue; if no, devise a second sampling phase to collect usable data.
2) Assess if bulk metals concentrations in surface water or sediments correlate with concentrations in vegetation, benthos, or fish. If yes, recommend going to step 4; if no, recommend step 3.
3) Assess if metals concentrations in sediment pore water correlate with vegetation or benthic community indices, or significant toxicity in invertebrates. If yes, to step 4, if no, no further action.
4) Assess if bioavailable metals concentrations in surface water or sediments correlate with concentrations in vegetation, benthos, or fish. If yes or no, go to step 5.
5) Assess if bioavailable metals concentrations in sediments correlate with significant toxicity in invertebrates. If yes, go to step 6; if no, no further action.
6) Assess if bioavailable metals concentrations in sediments correlate with vegetation community endpoints and benthic community indices. If yes, go to step 7; if no go to step 8.
7) Compare site data to reference site data and determine acceptable levels of bioavailable metals concentrations in sediments.
8) Compare site data to reference site data and determine acceptable levels of total metals concentrations in sediments.

Step 6: Specify Tolerable Limits on Decision Errors

Error margins are generally higher for sediment samples in comparison to water samples. Concentration variability of 35% or more between duplicate soil samples indicates that data should be used with caution.

Laboratory QC sample recoveries will be reviewed to determine if they are within acceptable limits. These recovery units are established by the analytical method. Matrix spikes and laboratory control samples will be conducted in accordance with the validation procedures.

The acceptable limits on decision errors should be smallest (i.e., have the lowest probability of error) for cases where one has the greatest concern for decision errors. This means that if one type of error is more serious than another, then its acceptable limits should be smaller (more restrictive). In addition, the limits on decision errors are usually highest (high probability of error can be tolerated) near the action level, since the consequences of decision errors are generally less severe as the action level is approached (EPA, 1994b). The acceptable limits of decision errors for this study will be that analytical results (the 95% upper confidence level [UCL] for surficial concentrations) must be exactly equal to or below background concentrations to be excluded from further action consideration.

Step 7: Optimize the Design for Obtaining Data

This step identifies a resource-effective data collection design for generating data that are expected to satisfy the DQOs. The data collection design (sampling program) is described in detail in the FSP, Part 1 of this SAP. Metal concentration data are not available with any reasonable density within the chosen areas. Given the relatively small land area under study (approximately 8.4 acres) the number of samples was selected based on spatial distribution.

5.5.2. Data Measurement Objectives

Based on the information provided on the DQOs, all samples will be analyzed using EPA methods and other standard analytical techniques. Every reasonable attempt will be made to obtain a complete set of usable analytical data. If a measurement cannot be obtained or is unusable for any reason, the effect of the missing data will be evaluated by the QAO and Site Manager. Table 4.1 summarizes the analytical methods and data measurement objectives for analyses that will be conducted in the field investigations.

5.6 Quality Assurance Guidance

The field QA program has been designed in accordance with EPA's *Guidance for the Data Quality Objectives Process* (EPA, 1994b), and the EPA's *Requirements for Quality Assurance Project Plans for Environmental Data Operations* (EPA, 1997).

5.6.1. Precision, Accuracy, Representativeness, Completeness, and Comparability Criteria

Precision, Accuracy, Representativeness, Completeness, and Comparability (PARCC) parameters are indicators of data quality. PARCC goals are established for the site characterization to aid in assessing data quality, as discussed in the following paragraphs:

Precision. The precision of a measurement is an expression of mutual agreement among individual measurements of the same property taken under prescribed similar conditions. Precision is quantitative and most often expressed in terms of relative percent difference (RPD). Precision of reported results is a function of inherent field-related variability plus laboratory analytical variability. Various measures of precision exist, depending upon "prescribed similar conditions." Field duplicate samples (1 duplicate / 20 samples) will be collected to provide a measure of the contribution to overall variability of field-related sources. Contribution of laboratory-related sources to overall variability is measured through various laboratory QC samples. The acceptable RPD limits for field duplicates are less than 35% for soil, water and sediments. Chemical analytical data will be validated for precision using field duplicates, laboratory duplicates, matrix spike/matrix spike duplicates (MS/MSDs), and laboratory control sample/laboratory control sample duplicates (LCS/LCSDs), as applicable.

Accuracy. Accuracy is the degree of agreement of a measurement with an accepted reference or true value, and is a measure of the bias in a system. Accuracy is quantitative and usually expressed as the percent recovery (%R) of a sample result. Ideally, it is desirable that the reported concentration equals the actual concentration present in the sample. Acceptable QC limits for %R are 75% to 125% for LCS/LCSDs, method-defined for surrogates, and laboratory-defined for MS/MSDs. Chemical analytical data will be validated for accuracy using surrogates, MS/MSDs, and LCS/LCSDs, as applicable.

Representativeness. Representativeness expresses the degree to which sample data accurately and precisely represent (a) a characteristic of a population, (b) parameter variations at a sampling point, and/or (c) an environmental condition. Representativeness is a qualitative parameter that is most concerned with the proper design of the sampling plan and the absence of cross-contamination. Good representativeness will be achieved through: (a) careful, informed selection of sampling sites, (b) selection of testing parameters and methods that adequately define and characterize the extent of possible contamination and meet the required parameter reporting limits, (c) proper gathering and handling of samples to avoid interference and prevent contamination and loss, and (d) collection of a sufficient number of samples to allow characterization. Representativeness is a consideration that will be employed during all sample location and collection efforts and will be assessed qualitatively by reviewing field procedures and reviewing actual sampling locations versus planned locations.

Completeness. Completeness is a measure of the amount of usable data obtained from a measurement system compared to the amount that was expected to be obtained under normal conditions. Evaluating the PARCC parameters will assess usability. Those data that are validated and need no qualification, or are qualified as estimated data, are considered usable. Rejected data are not considered usable. Completeness will be calculated following data evaluation. For this work, a completeness goal of 90% is projected for each analytical test. If this goal is not met, additional sampling may be necessary to adequately achieve project objectives.

Comparability. Consistency in the acquisition, handling, and analysis of samples is necessary for comparing results. Where appropriate, the results of analyses obtained will be compared with the results obtained in previous studies. Standard EPA analytical methods and QC will be used to ensure comparability of results with other analyses performed in a similar manner. Comparability is a qualitative parameter and cannot be assessed using QC samples.

5.6.1 Field Measurements

Field measurements specified in Table 4.1 will be conducted during the investigation. All procedures recommended by the manufacturer will be followed in calibrating and operating the instruments. Analytical methods, reporting limits, holding times, and QC analyses are discussed below.

5.7 Laboratory Analytical Methods

Analytical methods, with corresponding laboratory reporting limits (LRLs) are specified on Table 4.1. Laboratories with established protocols and quality assurance procedures that meet or exceed applicable EPA guidelines will analyze samples by following these methods. Samples will be analyzed using EPA-approved or recommended methods when available and will include all associated QA/QC procedures recommended in each method.

Samples will be submitted to American Environmental Consultants Laboratory (AEC) in Salt Lake City, Utah. AEC is certified with the State of Utah. Appendix B contains AEC's QA/QC manual, and certification letters from the Utah Department of Health and Division Bureau of Laboratory Improvement. If another lab performs analyses, it must meet the following criteria and submit all QA documentation to the EPA for approval as described above:

- Demonstrated ability to achieve the required detection limits,
- Certified by the State of Utah, and
- Established internal QA/QC program.

If contradictions between the laboratory QA/QC manuals or other documents are identified, information in this SAP supersedes all other documents.

For sediment samples, the laboratory shall assume that the entire sample submitted for analysis is representative material. To avoid substance losses, any overlying water in sediment samples received from the field will be mixed into the sample before removal of a subsample for analysis.

5.7.1. General Sediment and Water Analysis

5.7.1.1 Sediment

Sediment samples will be analyzed for general parameters, consisting of particle size distribution, nutrients (total phosphorus and total Kjeldahl nitrogen), total organic carbon (TOC), moisture content, and pH (Table 4.1). In addition, the sediment samples will be analyzed for metals including aluminum, silver, selenium, antimony, cadmium, lead, arsenic, zinc, manganese, chromium, copper, iron and mercury (Table 4.1).

Analysis for sediment particle size will follow Plumb (1981) with minor modifications. Each sediment sample will be separated into two particle-size fractions (>63 µm [sand] and <63 µm [silt and clay] only) using standard wet-sieve analyses. The material retained on the sieve will be rinsed into a pre-cleaned, tared beaker, dried at 90°C, and weighed. Percent of total dry weight for each of the two size fractions will be calculated. The weight of the sand fraction will be added to the weight of the silt/clay fraction and compared to the starting weight for each sample.

5.7.1.2 Surface Water and Sediment Porewater

Samples of surface water and sediment pore water will be analyzed for the following water quality parameters: alkalinity, conductivity, hardness and associated ions (calcium, magnesium, sodium, potassium, carbonate, and bicarbonate), chloride, nutrients (total phosphorus and total Kjeldahl nitrogen, ammonia, nitrite, and nitrate), sulfides, sulfate, total suspended solids (TSS), total dissolved solids (TDS), pH, dissolved organic carbon, and temperature (Table 4.1). The surface water and sediment porewater samples will also be analyzed for metals including aluminum, silver, selenium, antimony, cadmium, lead, arsenic, zinc, manganese, chromium (total and chromium VI), copper, iron and mercury (Table 4.1).

5.7.2 Bioavailable Metals Analysis

Bioavailable metals will be measured by analyzing surface water and sediment pore water samples following the methods specified in Table 4.1. Water samples will be analyzed for aluminum, antimony, arsenic, cadmium, chromium (total and chromium VI), copper, iron, lead, manganese, mercury, selenium, silver, and zinc, and for calcium, potassium, magnesium, and sodium.

Surface water samples will be analyzed for dissolved metals by filtering water samples through a 0.45 µm membrane filter in the field. Porewater samples will also be filtered after collection.

5.7.3 Total Metals

Sediment and organism tissue samples will be analyzed for total metals.

5.7.3.1 Bulk Sediment

Total metals will be measured in bulk sediment samples according to methods specified in Table 4.1. Sediment samples will be analyzed for aluminum, antimony, arsenic, cadmium, chromium, copper, iron, silver, manganese, lead, selenium, zinc, and total mercury. The samples will be analyzed by EPA method 6010B (ICP/AES) or by EPA method 8020 (ICP/MS), depending on the availability of laboratory instrumentation. Sample digestion will be completed by EPA method 3050B, using acids as appropriate for the determinative method.

5.7.4 Biological Tissue Analysis

Benthic macroinvertebrate, fish, and plant tissue samples will be digested and analyzed for the same metals as bottom sediments (Table 4.1). Metals in tissue samples will be analyzed by the same methods specified for sediments.

5.7.5 Sediment Toxicity Testing

Chronic toxicity of wetland and pond sediment to benthic organisms will be measured using EPA Test Methods (U.S. EPA 2000). The two tests use the amphipod *Hyalella azteca* in 28-day exposures and the midge *Chironomus tentans* as test species in 50-65 day exposures to bulk sediments. The test medium will be collected from the surface to 10 cm (i.e., the "biologically active zone") of sediment as described in Section 4.5.2.1 above. Both negative controls (i.e., clean sediment) and positive controls (i.e., reference toxicant) will be used to ensure that the test organisms are suitably healthy and responsive for testing.

Ten on-site sediment samples and two reference sediment samples will be submitted to the laboratory for toxicity testing. Endpoints for the tests are:

- *Hyalella* test
  - Percent survival on Day 28
  - Growth (as length) on Day 28
  - Reproduction (number of eggs per female) on Day 28.

- *Chironomus* test
  - Percent survival of larvae (Day 20 and end), pupae, and adults (end)
  - Growth (as length) on Day 20
  - Percent emergence of females and males
  - Number of egg cases
  - Reproduction (number of eggs per female)
  - Percent hatched eggs (potential endpoint)

QA/QC procedures for the toxicity tests include use of positive and negative controls and measurement of water quality conditions (i.e., conductivity, hardness, pH, alkalinity, ammonia, sulfide, temperature, and dissolved oxygen) in each test chamber during the 10-day exposure period. The positive controls use exposure to a reference toxicant such as cadmium chloride to ensure that the test organisms are suitably sensitive for testing (i.e., they respond to the reference toxicant in a dose-responsive manner and the LC50 values are within the expected range based on past studies). The negative controls use exposure to the culture medium to ensure that the test organisms are suitably healthy for testing (e.g., they are not overly stressed from handling, culturing, or other nontoxicant factors). Criteria for test acceptability will follow recommendations of U.S. EPA (2000)

5.7.6 Macroinvertebrate Community Analysis

Sorting and identification of aquatic macroinvertebrates will follow procedures in Aquatic Resources SOP, *Procedures for Benthic Macroinvertebrate Taxonomy and Enumeration* (To be provided by the laboratory conducting this analysis). Qualified taxonomists will make all taxonomic identifications, and a project reference collection will be established. Identifications will be made to the lowest practical taxonomic level. All samples will be archived for the duration of the project. The following information will be reported by the taxonomic laboratory to allow a complete review of the benthic data:

- The number of individuals of each taxon found in each sample
- The biomass (nearest 0.1 g dry weight) of each major taxonomic group in each sample
- The sorting efficiency for each sample and the identity of all samples that required sorting.

6.0 QUALITY CONTROL REQUIREMENTS

Quality control will include collecting field duplicates at a rate of 10 percent of the sample load for each sample type, and ensuring that the laboratory runs matrix spike/matrix spike duplicates at a rate of five percent of the sample load for each sample type. The field duplicates will be submitted "blind" to the sample laboratory, i.e., they will be given a separate sample identification number from the environmental sample, unidentifiable to the laboratory, as described above. Field duplicates will be run for the same analytical suite as the environmental samples.

Samples for preparation of matrix spikes and laboratory duplicates will be selected at random by the laboratory. Separate samples do not need to be collected in the field. The laboratory will perform and report all analyses under QA/QC procedures that include the results of method blanks, laboratory control samples, matrix spikes, and laboratory duplicates. Additional method-specific quality control procedures such as interference check samples, serial dilution, and internal standards will be used as specified for each analytical method in SW-846 (U.S. EPA 2003).

Due to the nature of the contaminants at this site, ambient, equipment and trip blanks will not be collected.

6.1 Instrument/Equipment Testing, Inspection, and Maintenance Requirements

All instruments and equipment will be regularly tested, inspected, and maintained according to manufacturers' instructions. Field equipment will be tested and inspected daily before use. Any equipment found to be not functioning properly will be repaired or replaced. Laboratory equipment will be tested, inspected and maintained in accordance with the laboratory QA/QC manual and manufacturers' recommendations.

6.2 Instrument Calibration & Frequency

6.2.1 Field Instruments

RMC will follow the manufacturer's specifications to calibrate any field equipment prior to each use. These manufacturers specifications are included in RMC's SOP's (Appendix A). A record of the calibration will be kept in the field logbook.

6.2.2 Laboratory Equipment

Procedures and schedules for the calibration of laboratory equipment are described in the appropriate SW-846 and EPA methods, and in the laboratory's Quality Assurance Plan. These procedures and schedules will be followed for all laboratory work.

6.2.3 Data Management

Data from AEC Laboratory will be submitted to United Park and RMC in both hard copy and electronic form. To avoid transcription errors, report tables will be prepared directly from the electronic submittals.

7.0 ASSESSMENT / OVERSIGHT

7.1 Assessments and Response Actions

This section describes the number, frequency, and type of assessment activities needed for this project. Assessments coordinated by the Project QA Officer will include: (1) a readiness review prior to initiating each major phase of field work; (2) surveillance during representative phases of the project; (3) a technical systems audit (TSA) conducted toward the end of the first week of field work; and (4) a data quality assessment (DQA).

The readiness review will be conducted with both the field staff and analytical laboratories as a technical check to determine if the staff, subcontractors, equipment, and record keeping systems are in place to start work in accordance with this QAPP. At the review, the QA Officer will review the project objectives, methodologies, record keeping requirements, and schedule with the field team and laboratories to make sure they are familiar and prepared to meet project requirements. The QA Officer will make sure all systems are ready before field work is initiated.

Surveillance will include weekly reviews of project progress and compliance with QAPP requirements. The project QA Officer will visit the field teams at the Site and observe their work habits and review project records. Based on the surveillance results, the QA Officer may propose corrective actions or changes to the field methods to the Project Manager.

A TSA will be conducted about halfway through the field portion of the project. The TSA is a thorough and systematic on-site qualitative audit where facilities, equipment, personnel, training, procedures, and record keeping and is conducted to determine conformance to the QAPP.

The DQA will be conducted to determine whether the data meet the assumptions that the DQOs and data collection design were developed under and whether the total error in the data are tolerable. This assessment activity will include complete data verification and validation as described in Section 5.0. *Guidance for the Data Quality Assessment Process* (EPA QA/G-9) will be consulted.

The QA Officer will report results of the assessment activities directly to the Project Manager who, with the assistance of the QA Officer, will be responsible for implementing any necessary corrective actions. The occurrence and resolution of major quality issues identified during assessment activities will be documented in memorandum to UPCM, the EPA Project Manager Jim Christiansen, and the UDEQ Project Manager Muhammad Slam.

8.0 DATA VALIDATION AND USABILITY

8.1 Data Review, Validation & Verification Requirements

The data validation process evaluates whether the specific requirements for an intended use have been fulfilled and ensures that the results conform to the users needs. The data validation process develops the QC acceptance criteria or performance criteria.

Data verification confirms that the requirements of the specified sampling and analytical methods were followed. This process involves reviewing the results of sampling and analysis to determine conformance with the QC requirements described for the project. The data verification process ensures the accuracy of data by using validated methods and protocols, and is often based on comparison with reference standards.

Requirements and methods for data validation and verification are listed in Tables 8.0 and 8.1.

8.2 Validation & Verification Methods

Data will be reviewed to ensure that the requirements stated in Table 4.1 and 8.0 were met. Data validation and verification will be conducted using the methods described in Table 8.1. Superfund's working definitions for data verification and validation are as follows:

> Data Validation: A consistent, systematic process that determines whether the data have been collected in accordance to the specification as listed in the approved QAPP. The process is independent of data validation and is conducted at various levels both internal and external to the data generator (laboratory).
>
> Data Validation: An evaluation of the technical usability of the verified data with respect to planned objectives. Data validation is performed external to the data generator (laboratory), using a defined set of performance criteria to a body of data in the evaluation process. This may include checks on some or all of the calculations in the data set and reconstruction of some or all final reported data from initial laboratory data (e.g., chromatograms, instrument printouts). It is in the data validation process that data qualifiers for each verified data are evaluated. It extends beyond the analytical method to protocols or QAPPs to address the overall technical usability of the generated data.

One hundred percent (100%) of the data will be validated according to Table 8.1 requirements by the Project QA Officer or a subcontractor experienced in conducting this type of data verification. Data will be reviewed as it is received, continuously throughout the project. If problems are uncovered as a result of the validation effort, the QA Officer and Project Manager will be immediately notified. The QA Officer or Project Manager will discuss possible corrective actions with the laboratory prior to implementation. The Project Manager will immediately notify EPA and UDEQ of any data verification or validation issues that may affect the success of the project.

Any deviations from the analytical control limits specified in Table 4.1 and 8.1 will be evaluated in terms of their effect on the data usability. Data usability will be assessed using the National Functional Guidelines for Data Review (Inorganic & Organic, February 1994). The completeness goal for the project is 90 percent valid data.

The results of the data validation and verification will be summarized in a Data Review Report, to be prepared after the completion of sampling and analysis activities at the site.

8.3 Reconciliation with Data Quality Objectives

The data validation and verification results will be compared to the DQOs stated in Table 5.0 and with the PARCC parameters described in Table 8.0. This evaluation will summarize the QA/QC performance by PARCC criteria including completeness calculations expressing the percent complete of valid data compared to the total number of samples collected. The result of the data validation and verification will be summarized in the Data Review Report described above.

8.4 Reporting Limits

The reporting limits provided in Table 4.1 are the minimum levels that the laboratory will report analytical results without a qualifier when an analyte is detected. The laboratory can typically detect analytes at concentrations of up to an order of magnitude lower than the reporting limits; in this case, when a positive detection is less than the reporting limit, the value may be reported and qualified as an estimated concentration.

8.5 Holding Times

Holding times are storage times allowed between sample collection and sample extraction or analysis (depending on whether the holding time is an extraction or analytical holding time) when the designated preservation and storage techniques are employed. Sample preservation and holding time requirements for samples collected in the field investigations are summarized in Table 4.1. Holding times for soil samples for analysis of metals is 180 days (30 days for mercury) with no preservative. Total organic matter, DOC, nutrients, and pH samples should be analyzed as soon as possible following collection. All samples will be cooled and stored at 4 degrees Celsius ±2 degrees Celsius until the requested analyses are performed.

8.6 Quality Control Analyses

To provide an external check of the quality of the field procedures and laboratory analyses, two types of QC samples will be collected and analyzed. Field replicate (duplicate) samples will be collected in order to distinguish between variability in results introduced by the field and sample handling prior to receipt by the laboratory and variability introduced by the laboratory procedures. These samples will be analyzed for metals. An equipment rinsate blank will be collected and analyzed for metals to assess potential contamination of sampling equipment for the analytes of interest. The collection and number of field QC samples that will be analyzed in this field program are discussed in Section 5.6 of this QAPP

In addition to the external QA/QC controls, the laboratory maintains internal QA procedures. Internal QC samples will include laboratory blanks (i.e., method blanks, preparation blanks), laboratory duplicates, MS/MSDs, and LCS/LCSDs, as discussed in Appendix B.

8.7 Special Training Requirements

The only special training required for this investigation is the health and safety training, as described in the RI HASP (RMC, 2001) for the project.

9.0 MEASUREMENT AND DATA ACQUISITION

This section covers sample process design, sampling methods requirements, handling and custody, analytical methods, QC, equipment maintenance, instrument calibration, supply acceptance, nondirect measurements, and data management.

9.1 Sample Process Design

The general goal of the field investigation is to verify and quantify the presence or absence of arsenic and metals in surface sediments. Sections 3.0 and 4.0 of this SAP describe the field sampling plan.

9.2 Sampling Methods Requirements

Sampling equipment, containers, and overall field management are described below.

9.2.1 Sampling Equipment and Preparation

Sampling equipment required for the field program for environmental sampling, health and safety monitoring, equipment and personal decontamination, and general field operations are presented in Table 4.0 of this SAP.

Field preparatory activities include review of SOPs, procurement of field equipment, laboratory coordination, confirmation of site access, as well as a field planning meeting attended by field personnel and QA staff. Site mobilization is described in Section 4.0 of this SAP.

9.2.2 Sample Containers

Sediment samples for laboratory analysis will be collected in 8-ounce wide-mouth glass jars. Plant tissue samples and sediment samples for toxicity tests will be collected in 1-gallon, self-sealing plastic bags. Surface water samples for metals analysis will be collected in 1-liter poly bottles. Containers for the environmental samples that will be collected during the field program are specified in Table 4.1.

9.2.3 Sample Collection

Samples collected during this field program will consist of surface sediment, surface water, benthic macroinvertebrates, fish, vegetation, and QC samples. All sample collection procedures are outlined in Section 4.5 and SOPs in Appendix A. The following SOPs apply to all applicable sample collection activities:

RMC SOP 1, Surface Water Sampling and General Water Sample Handling

RMC SOP 5, Sample Handling, Documentation and Shipping
RMC SOP 6, Sampling Equipment Decontamination
SRC-OGDEN SOP 01, Porewater Sampling from a Micro Push Point or Mini Piezometer
EPA/ERT #2037, Terrestrial Plant Community Sampling,
Procedures for Benthic Macroinvertebrate Taxonomy and Enumeration (To be provided by the laboratory conducting this analysis)
Exponent SOP BI-01, Decontamination of Equipment—Tissue
Exponent SOP BI-04, Fish Collection Procedures using an Electroshocker
Exponent SOP BI-05, Fish Collection Procedures using a Seine Net
Exponent SOP BI-08, Fish Processing Procedures
Exponent SOP BI-11, Aquatic Invertebrate Processing Procedures
Exponent SOP BI-12, Benthic Macroinvertebrate Sampling using a Grab Sampler
Exponent SOP BI-13, Vegetation Sampling
Exponent SOP SD-05, Surface Sediment Sampling using an Ekman Grab Sampler
Exponent SOP SD-10, Sediment Coring Using a Drive Rod Check-Valve Corer

9.3 Sample Handling and Custody Requirements

Custody and documentation for field and laboratory work are described below, followed by a discussion of corrections to documentation.

9.3.1. Field Sample Custody and Documentation

Samples for TAL metals submitted through the CLP will be labeled in accordance with the *Sampler's Guide to the Contract Laboratory Program* (EPA, 1990). Samples analyzed through laboratories coordinated by RMC will be labeled using procedures established in RMC SOP 5 (Sample Handling and Documentation). Sample labels will include the site name, sample identification number, and required analyses. Additional sample collection information including the date and time of sample collection, and sampler's initials will be recorded on the labels in permanent black ink markers or pens at the time of sample collection.

9.3.2. Chain-of-Custody Requirements

A Chain-of-Custody Record will be completed at the time of sample collection. An EPA Chain-of-Custody Record will be used for samples submitted for analysis through the CLP, and an SAIC Chain-of-Custody Record or one provided by the laboratory will be used for non-CLP samples. Field personnel will record the sample identification number, sampling date and time, sample matrix, sampler's initials, and analytical requirements in permanent black ink pens. Completed Chain-of Custody Records will be reviewed for completeness by the Field Operations Manager prior to sample shipment. Samples will be relinquished under the Chain-of-Custody Procedures identified in EPA's *Sampler's Guide to the Contract Laboratory Program* (1990) and RMC SOP 5 (Sample Handling and Documentation).

9.3.3. Sample Packaging and Shipping

Samples will be hand delivered to the laboratory.

After the sample containers are sufficiently packaged, the inner plastic bag lining the cooler will be sealed around the samples by twisting the top and securely taping the bag closed. Ice (sealed in bags) will be placed between the inner and outer plastic bags, with the latter taped and sealed closed. A temperature blank will be included with each cooler in order to record the cooler temperature upon receipt by the laboratory.

9.3.4. Field Logbooks and Records

Documentation of field activities will be conducted in accordance with RMC SOP 5 (Sample Handling and Documentation). The field sampling team will maintain a comprehensive field logbook that includes notes regarding instruments used, site and weather conditions, GPS coordinates, vegetative community observations, sample time, sampler's name, analytical parameters, and sample handling and chain of custody. The field activities will be recorded in bound, sequentially numbered, waterproof notebooks. All entries will be will be made in permanent black ink, will be clear, objective, and legible. Representative photographs will also be taken of field activities and sample locations, and a description will be recorded in the logbook. Photographs will be taken at each plant sampling location in the Phase II investigation. The Field Operations Manager is responsible for maintenance and document control of the field logbooks.

9.3.5. Laboratory Custody Procedures and Documentation

Laboratory custody procedures are provided in each laboratory's QA Manual. Upon receipt at the laboratory, each sample shipment will be inspected to assess the condition of the shipping cooler and the individual samples. This inspection will include measuring the temperature of the cooler (if cooling is required) to document that the temperature of the samples is within the acceptable criteria and verifying sample integrity. The enclosed chain-of-custody records will be cross-referenced with all of the samples in the shipment. Laboratory personnel will then sign these chain-of-custody records and copies provided to EPA Quality Assurance Coordinator will be placed in the project file. The sample custodian may continue the chain-of-custody record process by assigning a unique laboratory number to each sample on receipt. This number, if assigned, will identify the sample through all further handling. It is the laboratory's responsibility to maintain internal logbooks and records throughout sample preparation, analysis, data reporting, and disposal.

9.3.6. Corrections To and Deviations From Documentation

For the logbooks, a single strikeout initialed and dated is required for documentation changes. The correct information should be entered in close proximity to the erroneous entry. All deviations from the guiding documents will be recorded in the logbook(s).

9.4 Analytical Methods Requirements

Samples collected during this project will be analyzed in accordance with standard EPA and/or nationally-accepted analytical procedures. The selected EPA-approved laboratories will adhere to all applicable QC requirements established by the subcontract. The methods to be used for chemical analysis and the associated holding times are shown in Table 4.1.

9.5 Quality Control Requirements

Field, laboratory, and internal office QC are discussed below.

9.5.1. Field Quality Control Samples

Quality control checks will be employed during field activities to ensure the quality and integrity of sample collection. Both field duplicate and equipment rinsate QC samples will be collected in the field and shipped to the appropriate laboratory for analysis, as described in Section 5.10.

All field duplicates will be collected as close as possible to the same point in time and space as the primary field sample. Field duplicate sample will be prepared at a frequency of 20 percent of all sediment samples obtained during the study, and will be handled and analyzed in the same manner as the environmental samples.

9.5.2. Laboratory Quality Control Samples

The approved EPA contract laboratory(ies) will follow all laboratory QC checks, as defined in the analytical methods listed in Section 5.6. Quality control data are necessary to determine precision and accuracy and to demonstrate the absence of interferences and/or contamination. Each type of laboratory-based QC will be analyzed at a rate of 5 percent or one per batch (a batch is a group of up to 20 samples analyzed together), whichever is more frequent. Results of the QC will be included in the QC package and QC samples may consist of laboratory blanks, laboratory duplicates, MS/MSDs, and/or LCS/LCSDs, whichever are applicable, and any other method-required QC samples.

Blank samples will be analyzed to assess possible contamination so that corrective measures may be taken, if necessary. Duplicate samples are aliquots of a single sample that are split on arrival at the laboratory or upon analysis. Results obtained for two replicates that are split in a controlled laboratory environment may be used to assess laboratory precision of the analysis. MS/MSD and LCS/LCSD analyses may be used to determine both precision and accuracy.

Both normal and QC samples will be spiked with surrogate compounds, when applicable, and a percent recovery will be calculated for each surrogate.

9.5.3. Internal Quality Control Checks

Internal QC checks will be conducted throughout the project to evaluate the performance of the project team during data generation. All internal QC will be conducted in accordance with EPA CLP methods and requirements.

9.6 Equipment Maintenance Procedures

All laboratory equipment will be maintained in accordance with each laboratory's SOPs.

9.7 Instrument Calibration Procedures and Frequency

Calibration of field and laboratory instruments is addressed in the following subsections.

9.7.1. Field Equipment

Field instruments used in the field investigation consist of GPS units used to measure sample station coordinates and pH meters used to measure water samples. The GPS receivers require no special calibration procedure, and all measurements will be conducted according to the manufacturer's suggested procedures. There are few areas with overhead cover in the study area, and little difficulty is expected in acquiring adequate satellite signals.

Calibration of the pH meter will be performed prior to use in the field on at least a daily basis. In all cases, the pH meter will be calibrated and operated according to instructions supplied with the instrument, and calibration information will be recorded in the field log or instrument log. Solutions used for the calibration of pH meters will be within the expiration date supplied on the bottle label.

9.7.2. Laboratory Equipment

Calibration of laboratory equipment will be based on written procedures approved by laboratory management. Instruments and equipment will be initially calibrated and subsequently continuously calibrated at approved intervals, as specified by either the manufacturer or more updated requirements (e.g., methodology requirements). Calibration standards used as reference standards will be traceable to the EPA, National Institute of Standards and Technology, or another nationally-recognized reference standard source.

Records of initial calibration, continuing calibration and verification, repair, and replacement will be filed and maintained by the laboratory. Calibration records will be filed and maintained at the laboratory location where the work is performed and may be required to be included in data reporting packages.

9.8 Acceptance Requirements for Supplies

Prior to acceptance, all supplies and consumables will be inspected to ensure that they are in satisfactory condition and free of defects.

9.9 Non-Direct Measurement Data Acquisition Requirements

Non-direct measurement data include information from site reconnaissances, literature searches, and interviews. The acceptance criteria for such data include a review by someone other than the author. Any measurement data included in information obtained from the above-referenced sources will determine further action at the Richardson Flat site only to the extent that those data can be verified.

9.10 Data Management

Sample results and QC data will be delivered to the EPA RPM as an electronic data deliverable (EDD) in addition to a hard-copied data package. Electronic copies of all project deliverables, including graphics, are maintained by project number. Electronic files are routinely backed up and archived.

10.0 REFERENCES

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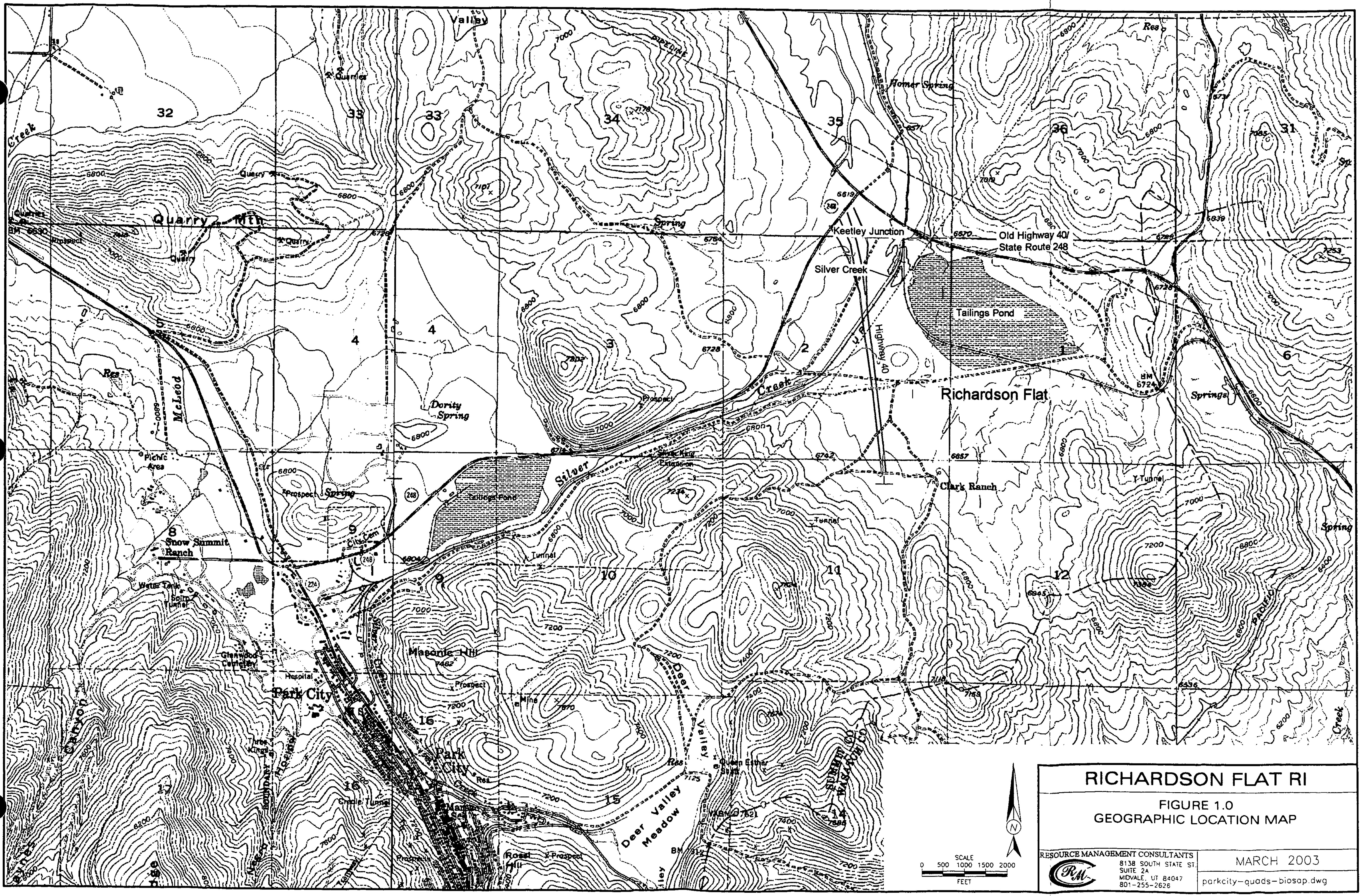


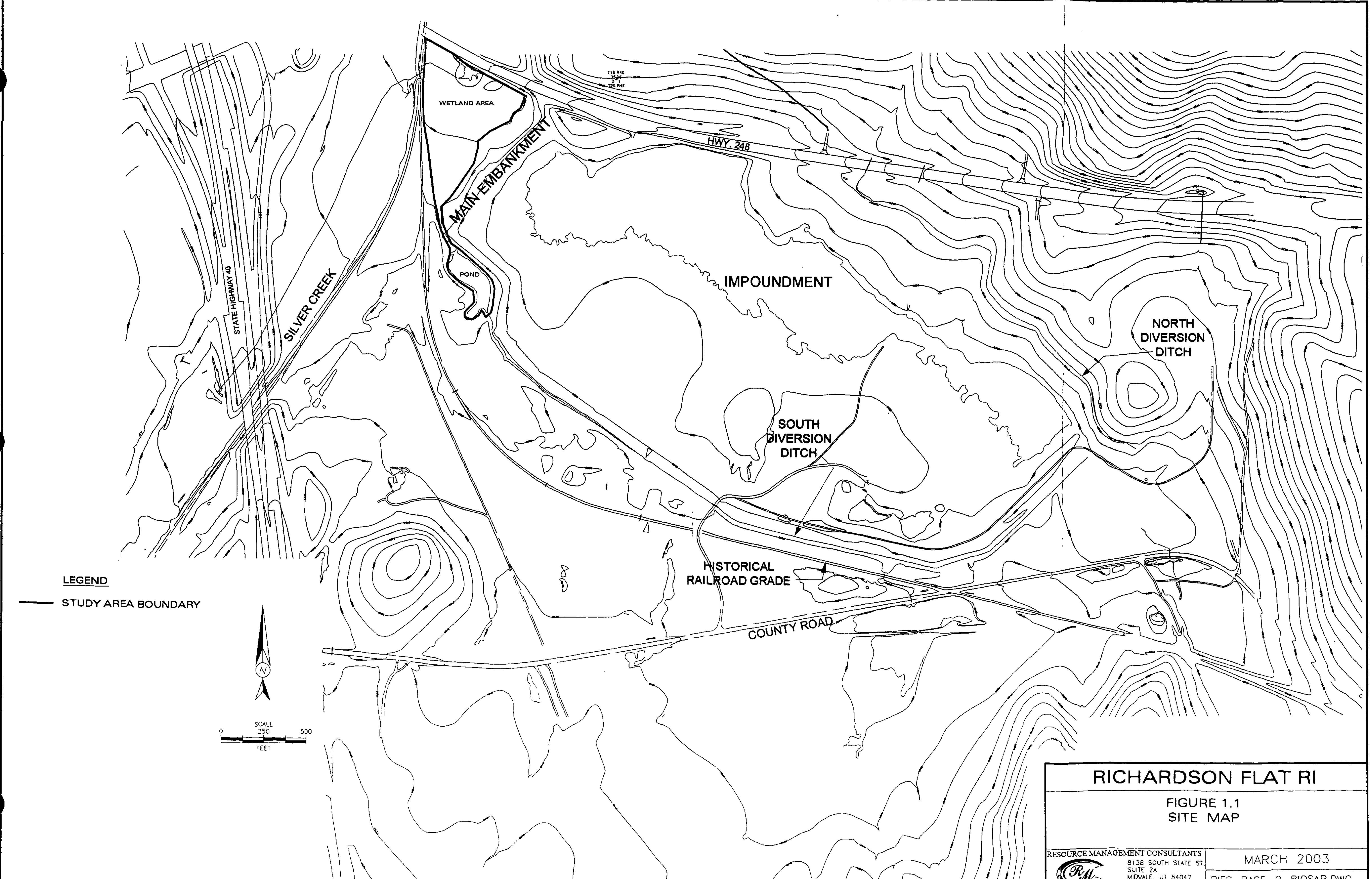
RICHARDSON FLAT RI

FIGURE 1.0
GEOGRAPHIC LOCATION MAP

RESOURCE MANAGEMENT CONSULTANTS
8138 SOUTH STATE ST.
SUITE 2A
MIDVALE, UT 84047
801-255-2626

MARCH 2003

parkcity-quads-biosap.dwg



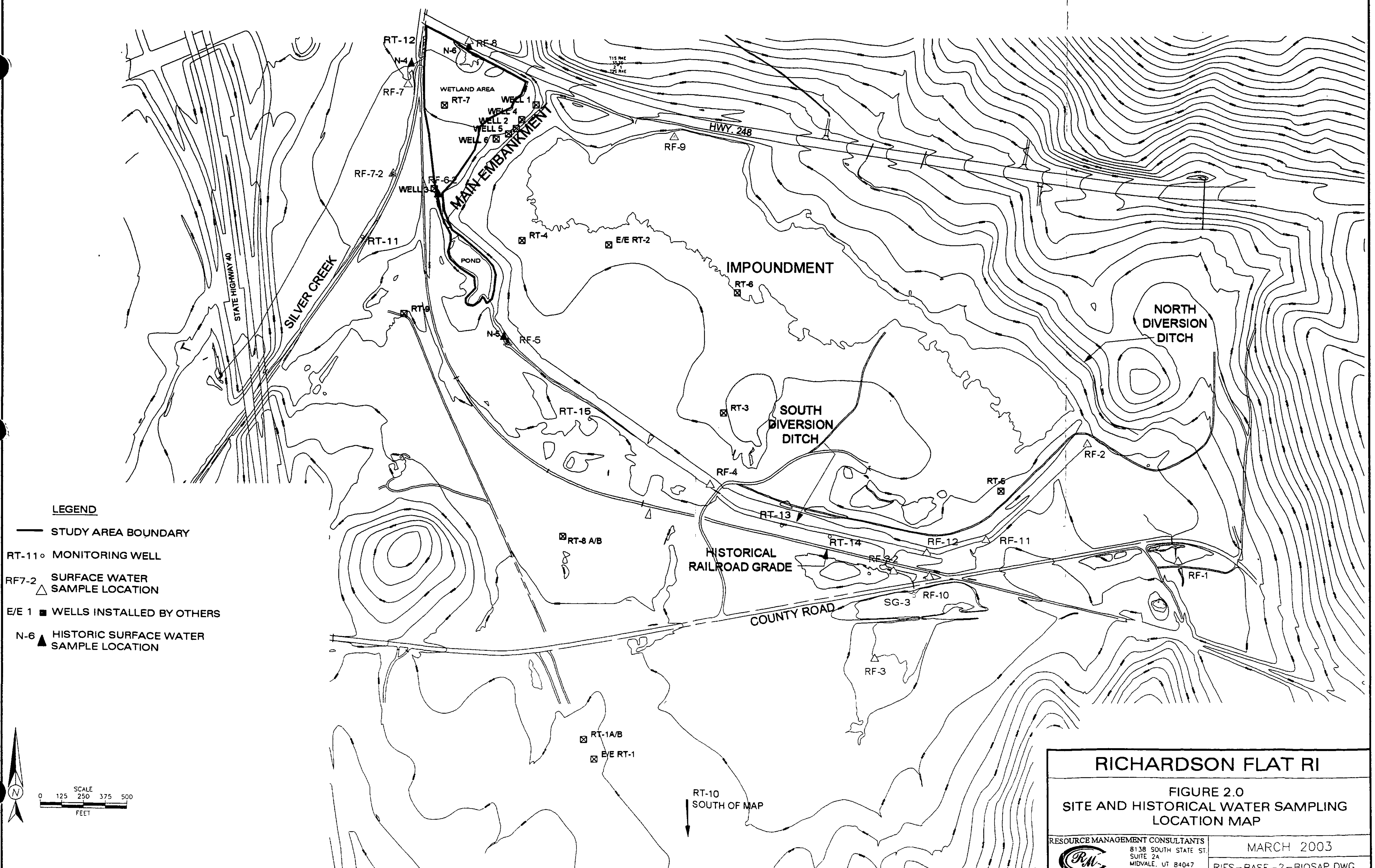
RICHARDSON FLAT RI

FIGURE 1.1
SITE MAP

RESOURCE MANAGEMENT CONSULTANTS
8138 SOUTH STATE ST.
SUITE 2A
MIDVALE, UT 84047
801-255-2626

MARCH 2003

RIFS-BASE-2-BIOSAP.DWG



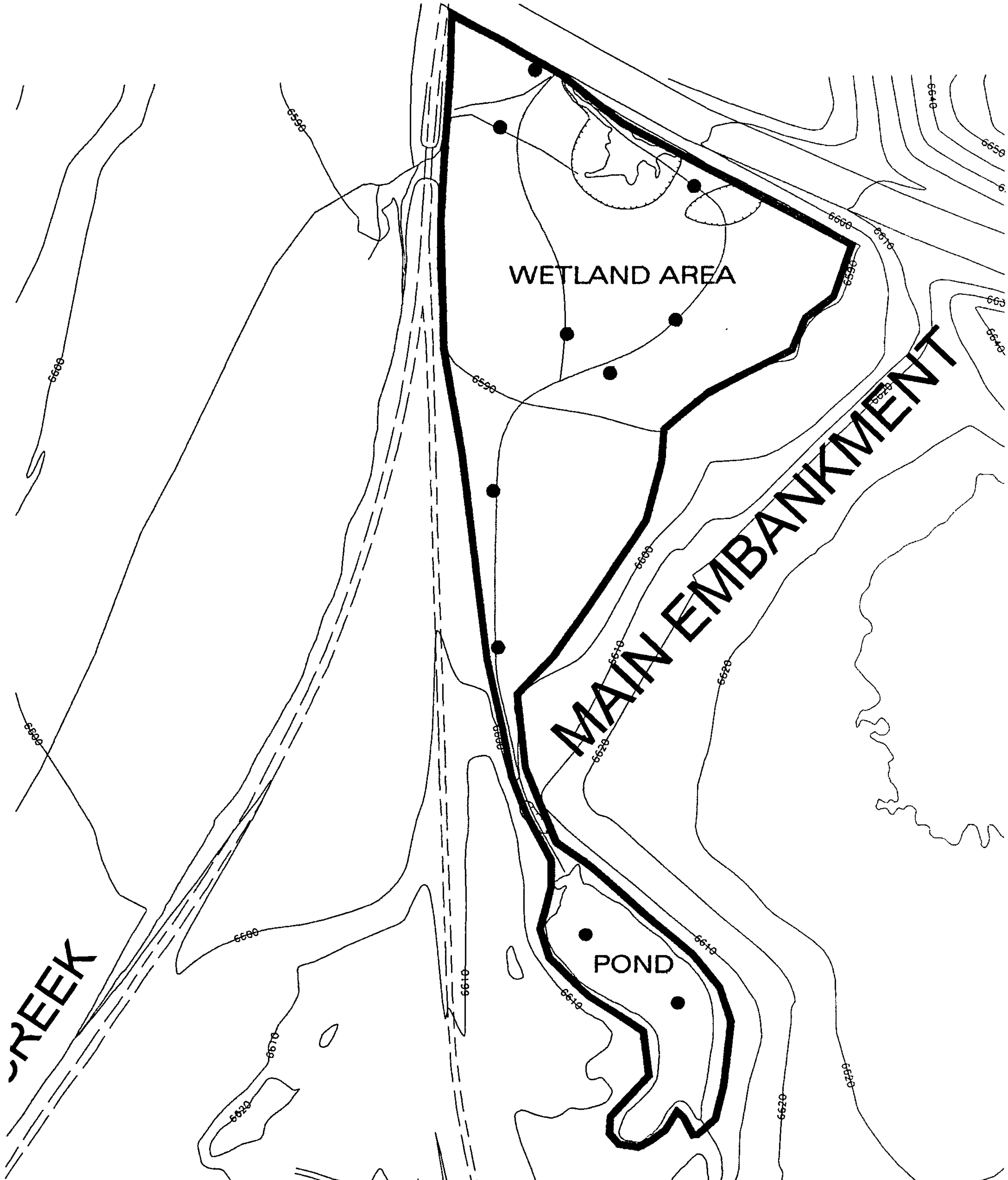
RICHARDSON FLAT RI

FIGURE 2.0
SITE AND HISTORICAL WATER SAMPLING LOCATION MAP

RESOURCE MANAGEMENT CONSULTANTS
8138 SOUTH STATE ST.
SUITE 2A
MIDVALE, UT 84047
801-255-2626

MARCH 2003

RIFS-BASE-2-BIOSAP.DWG



LEGEND

● ECOLOGICAL SAMPLE LOCATION

ECOLOGICAL STUDY AREA BOUNDARY

N

SCALE
0 50 100 150 200
FEET

RICHARDSON FLAT RI

FIGURE 3.0
SAMPLE LOCATION MAP

RESOURCE MANAGEMENT CONSULTANTS
8138 SOUTH STATE ST.
SUITE 2A
MIDVALE, UT 84047
801-255-2626

MARCH 2003

RIFS-BASE-2-BIOSAP.DWG

FIGURE 5.0 - Richardson Flat RI/FS Organizational Chart

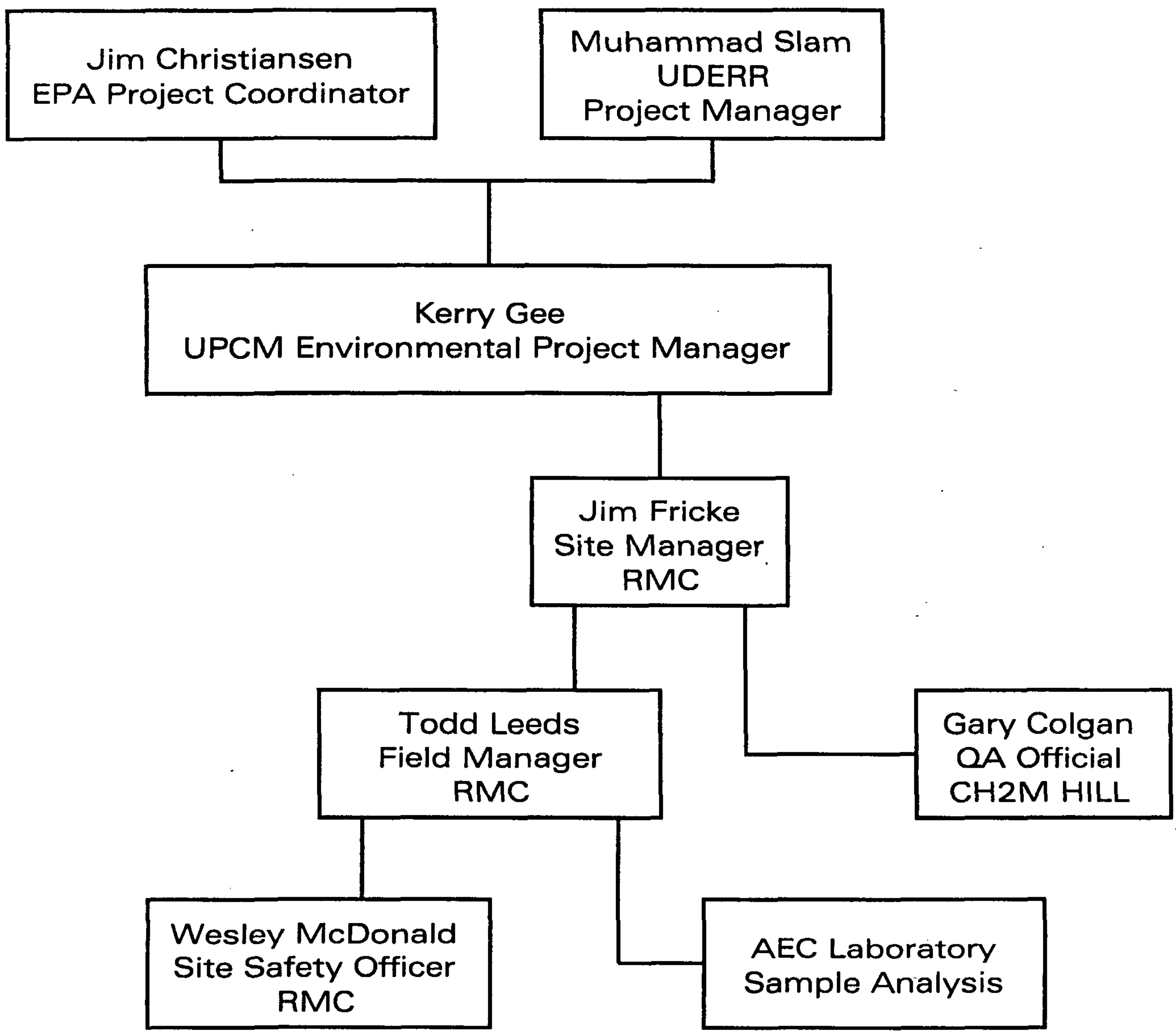


Table 4.0
Field Equipment and Supplies
Richardson Flat
Sampling and Analysis Plan

Sampling	Health & Safety	Decontamination	General
Stainless steel spoons (10)	Latex gloves (500 pr)	Plastic squirt bottles (2)	GPS
Steel shovels (2)	Sunscreen	Plastic trash bags (1 boxes)	Wooden stakes (20)
Stainless steel bowls (10)	Rubber boots	Deionized water (5 gallons)	Flagging (2 rolls)
Pruning shears (1)	Copy of HSAP	Nitric acid (10% solution - 1 gallons)	Coolers (2)
Stainless steel scissors (2)		Alconox (1 carton)	Copy of SAP
Self-sealing plastic bags (30 qt. size; 50 gal. size)		Plastic buckets (3 5-gal)	1 square meter PVC grid
Paper grocery bags (12 for biomass samples)		Scrub brushes (3)	
Field Logbook		Sprayer (1-liter)	
Plastic buckets (3 5-gal)			
Plastic trash bags (1 box of large - 30 count)			
Peristaltic pump and tubing			
0.45 um filters (25)			
Polyethylene bottles (30 1 liter, 60 0.5 liter)			
HNO3, H2SO4			

TABLE 4.1
Sample Collection Guide - Target Analytes and Collection Requirements
Richardson Flat
Sample and Analysis Plan

SURFACE WATER & SEDIMENT POREWATER

Parameters[1]	Method	LRL[2]	Container	Volume[4]	Temperature[6]	Preservative	Hold Days
DO	Field	NA	NA	NA	NA	NA	NA
pH, Temperature, Conductivity	EPA 150.1, 170.1, 120.1	NA	Polyethylene	Bottle 5	NA	None	1
Ag,As,Cu,Mn,Pb,Sb, (Dissolved)	SW-846 6010B or 6020	0.005	Polyethylene	Bottle 1,2	4°C	2 ml HNO_3 (ph<2)	180
Cd (Dissolved)	SW-846 6010B or 6020	0.001	Polyethylene	Bottle 1,2	4°C	2 ml HNO_3 (ph<2)	180
Se (Dissolved)	SW-846 6010B or 6020	0.004	Polyethylene	Bottle 1,2	4°C	2 ml HNO_3 (ph<2)	180
Cr (Dissolved)	SW-846 6010B or 6020	0.01	Polyethylene	Bottle 1,2	4°C	2 ml HNO_3 (ph<2)	180
Cr(VI) (Dissolved)	EPA 218.6	0.0004	Polyethylene	Bottle 1,2	4°C	2 ml HNO_3 (ph<2)	180
Al (Dissolved)	SW-846 6010B or 6020	0.05	Polyethylene	Bottle 1,2	4°C	2 ml HNO_3 (ph<2)	180
Zn (Dissolved)	SW-846 6010B or 6020	0.01	Polyethylene	Bottle 1,2	4°C	2 ml HNO_3 (ph<2)	180
Fe (Dissolved)	SW-846 6010B or 6020	0.1	Polyethylene	Bottle 1,2	4°C	2 ml HNO_3 (ph<2)	180
Hg (Dissolved)	EPA 245.1	0.0002	Polyethylene	Bottle 1,2	4°C	2 ml HNO_3 (ph<2)	28
Ca, K, Mg, Na	SW-846 6010B or 6020	2	Polyethylene	Bottle 2	4°C	None	180
Cl	EPA 325.2	1	Polyethylene	Bottle 3	4°C	None	28
NO_3, NO_2	EPA 353.2	0.1	Polyethylene	Bottle 4	4°C	H_2SO_4	28
CO_3, HCO_3	EPA 310.1	1	Polyethylene	Bottle 3	4°C	None	14
NH_3	EPA 350.1	0.1	Polyethylene	Bottle 4	4°C	H_2SO_4	28
Total P, Kjeldahl N	EPA 365.4 and 351.2	0.1	Polyethylene	Bottle 4	4°C	H_2SO_4	28
SO_4	SW-846 9036	2	Polyethylene	Bottle 3	4°C	None	28
Total Sulfides	EPA 376.2	2	Polyethylene	Bottle 6	4°C	ZnAc; NaOH (pH>9)	7
DOC	EPA 415.1	0.5	Polyethylene	Bottle 4	4°C	H_2SO_4	28
Alkalinity	EPA 310.1	1	Polyethylene	Bottle 3	4°C	None	14
Hardness	2340B[5] (calculation)	N/A	Polyethylene	Bottle 3	4°C	None	180
Cation/Anion Balance	Calculation	N/A	Polyethylene	N/A	4°C	None	NA
TSS	EPA 160.2	1	Polyethylene	Bottle 3	4°C	None	7
TDS	EPA 160.1	10	Polyethylene	Bottle 3	4°C	None	7

SEDIMENT - BULK

Parameters[1]	Method	LRL[3]	Container	Volume[4]	Temperature[6]	Preservative	Hold Days
pH	EPA 9045C	0.01	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	28
Al	SW-846 6010B or 6020	2 ppm	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Se, Sb, Cd, Pb, Ag, Mn	SW-846 6010B or 6020	0.05	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
As, Zn	SW-846 6010B or 6020	0.5	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Cr	SW-846 6010B or 6020	0.2	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Cu	SW-846 6010B or 6020	0.1	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Fe	SW-846 6010B	4	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Hg	SW-846 7471	0.02 ppm	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	28
Total P, Kjeldahl N	EPA 365.4 and 351.2	10	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Total Sulfides	EPA 9030	20	Glass Jar	4 oz.	4°C	No headspace	7
TOC	EPA Method 440.0	0.0005	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	28
Moisture Content	Gravimetric	0.10%	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	28
Particle Size Distribution	PSEP	0.10%	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180

SEDIMENT TOXICITY

Hyalella 28-day Survival, Growth, and Reproduction	EPA 2000	0	Glass	1 L (TFE lid liner)	4°C	N/A	14
Chironomus 20+-day Survival, Growth, and Reproduction	EPA 2000	0	Glass	1 L (TFE lid liner)	4°C	N/A	14

PLANT, MACROINVERTEBRATE, AND FISH TISSUE

Parameters[1]	Method	LRL[7]	Container	Volume[4]	Temperature[6]	Preservative	Hold Days
As, Cr, Fe, Zn, Cd, Mn	SW-846 6010B or 6020	0.5	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Al	SW-846 6010B or 6020	1	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Ag, Pb	SW-846 6010B or 6020	0.02	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Cu	SW-846 6010B or 6020	0.1	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Sb	SW-846 6010B or 6020	0.05	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Se	SW-846 7740	1	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	180
Hg	SW-846 7471	0.02 ppm	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	28
Moisture Content	EPA 160.3	0.10%	Glass Jar or Plastic Bag	4 oz.	4°C	N/A	28

BIOLOGICAL COMMUNITY

Parameters[1]	Method	LRL	Container	Volume[4]	Temperature[6]	Preservative	Hold Days
Benthic Macroinvertebrate - Species abundances	Aquatic Resources SOP	LPT	Polyethylene	1 L	ambient	formalin/ethanol	unlimited
Benthic Macroinvertebrate - Species richness	Aquatic Resources SOP	0	Polyethylene	1 L	ambient	formalin/ethanol	unlimited
Wetland Plant - Species abundances	Field Survey	LPT	N/A	N/A	N/A	N/A	N/A
Wetland Plant - Species richness	Field Survey	0	N/A	N/A	N/A	N/A	N/A

N/A - Not Applicable
LRL - Laboratory Reporting Limit
LPT - Lowest Practical Taxon
pH, Conductivity, Temperature, Flow
[1] Field Data Collected for each sample station/event includes:
[2] All units in mg/l except as noted.
3 All units are Parts Per Million (ppm) based upon dry weight unless otherwise noted.
[4] Laboratory analysis for the above parameters will require collection of the following sample volumes/preservation at each sample station:
[5] Standard Methods, 20th edition (APHA, 1989)
[6] Laboratory will measure the temperature of each cooler upon receipt to ensure proper temperature was maintained (4°C +/- 2°C)
[7] All units are Parts Per Million (ppm) based upon wet weight

Bottle 1 - 500 ml bottle filtered to 0.45μm and preserved with 2 ml HNO_3
Bottle 2 - 500 ml bottle unfiltered and preserved with 2 ml HNO_3
Bottle 3 - 1000 ml bottle unfiltered and unpreserved
Bottle 4 - 500 ml bottle unfiltered and preserved with 2 ml H_2SO_4
Bottle 5 - 500 ml bottle unfiltered and unpreserved for field parameters.
Bottle 6 - 500 ml bottle unfiltered and preserved with 2 ml zinc acetate and NaOH to pH

Table 5.0
Data Quality Objectives, Data Uses, Data Type, and QC levels
Richardson Flat
Sampling and Analysis Plan

Data Quality Objectives	Conceptual Site Model Exposure Pathway or Other Evaluation	Existing Data Summary	Data Needs	Data Use	Analysis Type	QC Level
Surface water sampling	Contact with surface water	Limited water quality data for diversion ditch and Silver Creek	Obtain surface water samples from wetland, pond, and reference areas	Evaluate wetland processes that remove metals from water; metals bioavailability; and exposure and risk to aquatic and wildlife receptors.	Water quality analysis, field parameters, dissolved metals	Definitive
Sediment porewater sampling	Contact with water	None	Obtain porewater samples from wetland, pond, and reference areas	Evaluate wetland processes that remove metals from water, metals bioavailability, relationship between bulk metals concentrations in sediments and pore water, and benthic macroinvertebrate exposure and risk.	Water quality analysis, field parameters, dissolved metals	Definitive
Sediment sampling (for chemical analyses)	Contact with sediments	Limited metals data for sediments in wetland	Obtain sediment samples from wetland, pond, and reference areas	Evaluate exposure and risk to aquatic receptors, wildlife, and plants.	Sediment metals analysis (dry weight), compositional properties of surficial sediments	Definitive
Sediment sampling (for bioassay)	Contact with sediments	None	Obtain sediment samples from wetland, pond, and reference areas	Evaluate exposure-response relationships and risk for aquatic macroinvertebrates.	*Hyalella* 28-d and *Chironomus* 20+-d survival, growth, and reproduction	Definitive
Plant tissue collection	Uptake by food chain	None	Obtain samples of plant tissue from wetland, pond, and reference areas	Evaluate uptake of metals by plants, assess wildlife exposure and risk.	Metals analysis of plants	Definitive
Benthic macroinvertebrate tissue collection	Uptake by food chain	None	Obtain samples of benthic macroinvertebrates from wetland, pond, and reference areas	Evaluate uptake of metals by aquatic macroinvertebrates; assess exposure and risk for aquatic receptors and wildlife .	Metals analysis of macroinvertebrates	Definitive

TABLE 8.0
Precison, Accuracy, Representativeness, Comparability and Completeness (PARCC)
Richardson Flat
Sample and Analysis Plan

Parameter	QC Program	Evaluation Criteria	Acceptance Criteria	Recommended Corrective Actions
Precision	Field Duplicate	Relative Percent Difference (RPD)	RPDs: soil, sediment and water samples +/- 35 percent if > 5 times LRL, or, +/- LRL if < 5 times LRL	Verify the RPD calculation. If correct, determine if matrix interference or heterogeneous samples are factors in poor RPD. If matrix effects or heterogeneous samples are not observed, reanalyze the associated investigative samples and MS/MSD. If appropriate, reextract or redigest and reanalyze the associated investigative samples and MS/MSD.
	Matrix Spike/Matrix Spike Duplicate (MS/MSD)	Relative Percent Difference (RPD)	See method-specific control limits[1]	Verify the RPD calculation. If this is correct, determine if matrix interference or heterogeneous samples are factors in poor RPD. If matrix effects or heterogeneous samples are not observed, reanalyze the method duplicate and associated investigative samples.
Accuracy	Matrix Spike (MS)	Percent Recovery	See method-specific control limits[1]	Verify the matrix spike percent recovery calculations and evaluate the LCS percent recovies. If the calculations are correct and the LCS recoveries are acceptable, determine if matrix interference is a factor in the poor recoveries. If matrix effects not observed, reanalyze the MS and associated samples. If appropriate, reextract or redigest and reanalyze the MS and associated investigative samples.
	Matrix Spike Duplicate (MSD)	Percent Recovery	See method-specific control limits[1]	Same as above.
	Laboratory Control Samples (LCS)	Percent Recovery	See method-specific control limits[1]	Verify the percent recovery calculations. Evaluate the standard to determine if it is faulty. If it is, prepare a new standard and reanalyze the LCS and associated investigative samples. If necessary, recalibrate the instrument. Do not continue analysis until problem solved.
Representativeness	Holding Times	Representative of Environmental Conditions	Holding Times Met 100 Percent	Evaluate whether data is critical to decision making. If so, resample and reanalyze for parameter exceeding holding time.
	Method Blanks	Qualitative Degree of Confidence	See method specific requirements[1]	Evaluate instrument, locate source of contamination, perform system blanks to confirm that system blanks meet performance criteria. Re-analyze method blank and associated samples. If method blank still above acceptance criteria, reextract or redigest the method blank and all associated samples.
	Equipment/Rinsate Blanks	Qualitative Degree of Confidence	Target analytes <1 X LRL; 5-10 X LRL for laboratory-inducted contaminants.	Suggests field sampling-induced contamination may have occurred. Evaluate all associated QC samples. If all other QC samples are within prescribed acceptance limits, but equipment blank is not (e.g., positive identification of target analytes observed), contact USEPA immediately to determine if resampling and/or reanalysis required.
	Field Duplicates	Qualitative Degree of Confidence	90 Percent of Field Duplicates Meet RPD Goals	If acceptance criteria not met, evaluate reasons for not meeting criteria (i.e., matrix interferences or heterogeneous samples) and make recommendations on whether resampling and/or reanalysis is necessary to improve degree of confidence.
Comparability	Standard Units of Measure	Qualitative Degree of Confidence	Laboratory Methods Followed	Revise analytical reports with correct units.
	Standard Analytical Methods		SOPs Followed	If SOPs not followed, evaluate whether reanalysis is necessary to obtain reliable data.
Competeness	Complete Sampling	100 Percent Valid[2] Samples	90 Percent Valid[2] Data	If not enough samples were collected for project needs, collect and analyze additional samples for parameters needed for key decisions.

[1] Laboratory Control limits are specific to individual analytical/digestion methods and any deviation outside control limits are reported (see method-specific SOPs in Appendix A).
[2] Valid means that samples meet all evaluation criteria (i.e., are not rejected for any reason).

Precision is a measure of how repeatable data are and is often measured by sample duplicates.
Accuracy is a measure of how close the data are to the actual, or real value, measured by certified reference materials and matrix spikes.
Representativeness is a measure of how representative a sample is of the sample population and is achieved by accurate sampling procedures and appropriate sample homogenization.
Comparability looks at ongoing projects and how variable one set of data is relative to another. Comparability helps to measure the scientific consistency of the system to past work.
Completeness is a measure of how many data points collected are usable; 90% usable data is considered to be an acceptable value for completeness.

TABLE 8.1
Data Validation and Verificaiton Requirements
Richardson Flat
Sample and Analysis Plan

Data Validation and Verification Steps		Data Validation and Verification Methods
Samples were collected according to established locations and frequencies.	→	Comparison with Sampling Plan
Sample collection and handling followed established procedures.	→	Review of field notes, field procedures and COCs
Appropriate analytical methods were used; internal laboratory calibration checks were performed according to the method-specified protocol.	→	Review of analytical methods and case narratives provided with laboratory reports. Documentation of any communications with laboratory concerning problems or corrective actions.
Required holding times and laboratory reporting limits were met.	→	Comparison with established holding times and LRLs.
Field Duplicates for QA/AC	→	Field duplicates met acceptance criteria tabulation of RPDs and comparison with PARCC parameters
Acceptance criteria (see Table 8.0) for field and laboratory QC samples (field blanks, field dups, equipment/rinsate blanks, method blanks, LCS) were met.	→	Tabulation of RPDs and spike recoveries, and direct comparison with method-specific acceptance criteria (see SOPs in Appendix A). Comparison with PARCC parameters.
Appropriate steps were taken to ensure the accuracy of data reduction, including reducing data transfer errors in the preparation of summary data tables and maps.	→	Maintain permanent file for laboratory hardcopies of analysis reports. Minimize retyping of data and error check data entered into database, tables, maps, etc.

RPD = Relative Percent Difference
LRL = Laboratory Reporting Limit

APPENDIX A

STANDARD OPERATING PROCEDURES

RMC SOP 1
STANDARD PROCEDURES FOR COLLECTION OF SURFACE WATER SAMPLES AND GENERAL WATER SAMPLE HANDLING

1.0 Purpose

This SOP describes the procedures that will be used for collection of surface water samples. The procedures will ensure that samples are collected and handled properly and that appropriate documentation is completed. The procedures outlined in this SOP detail the procedures used for the treatment/handling of all water samples collected.

2.0 Sampling Equipment

- Field data sheets / Field notebook / Chain of Custody Forms (COC) – Documentation of sample activities, field notes, sample custody and analyte list for laboratory.
- Sample containers – Containers provided by laboratory for the collection, storage and transportation of samples.
- Direct reading instruments – field instruments to measure pH, conductivity and temperature.
- Disposable sampling gloves – to prevent exposure to water and the prevention of cross-contamination.
- Custody seals – seals to be placed on sample containers to maintain sample integrity.
- 0.45 um filter apparatus with inert filters – for filtering samples in preparation for the analysis of dissolved metals.
- Nitric acid (HNO_3, supplied by the analytical laboratory) – for sample preservation.
- Water velocity meter and tape measure – to measure stream flow (where applicable).
- Distilled water – for rinsing direct reading instruments.
- Custody seals – seals to be placed on sample containers to maintain sample integrity.

3.0 Procedure

Sample bottles will remain sealed until the water sample is collected. At that time, the bottle lid will be removed and placed, top down, in an appropriate place. The sample bottle will be placed under the flow of water. The container will be rinsed three times prior to sample collection. If wading is required for sample collection, the sample must be collected upstream of wading personnel to avoid the sampling of suspended sediments. After rinsing, the sample container will be completely filled; any overflow of the sample container will be kept to a minimum. Sediment disturbance shall be kept to an absolute minimum. The sample cap will then be replaced on the sample bottle. All surface water samples will be collected in accordance with containers, volumes, preservatives, temperatures and holding times as outlined in Table 4.1 of the Sampling and Analysis Plan.

4.0 Dissolved Metals Analysis

Surface water samples collected for analysis of Dissolved (D) Metals will be a minimum volume of 500 ml, collected in a poly or glass container. The samples will be field filtered. The field filtering methodology will include the following steps:

1: Sample shall be collected in a 1000 ml bottle.

2: Sample is poured into the top of the disposable plastic filter.

3: Vacuum pump is attached to the filter and pumped.

4: When the bottom compartment of the filter is full, the water is to be transferred into a 500 ml sample container which shall be rinsed three times, the sample will be preserved with 2 ml of nitric acid (HNO_3), sufficient to bring the sample to pH <2.

5: The pH level in samples will be verified using pH paper before bottles are sealed.

5.0 Total Metals Analysis

Surface water samples collected for analysis of Total (T) Metals will be a minimum volume of 500 ml, collected in a poly or glass container, and preserved with 2 ml of nitric acid (HNO_3), sufficient to bring the sample to pH <2. The pH level in samples will be verified using pH paper before bottles are sealed.

6.0 Cations/Anions and Total Suspended Solids

Cations/Anions and Total Suspended Solids samples shall be collected in accordance with the methodologies outlined in the Procedure section of this SOP. Samples will not be preserved.

7.0 Field Parameter Collection – pH, Temperature and Conductivity

Temperature, pH and Conductivity measurements will be collected in the field at the time of sampling. All Direct reading field instruments will be calibrated prior to daily use according to the manufacturers specification manual/instructions provided with each instrument. Field data will be collected according to the manufacturers specification manual/instructions for each instrument. Water samples shall be placed into a clean container such as a sample bottle or 1 gallon bucket. The instrument probes will be placed into the water sample. Field data collected in this procedure will be recorded in the field notebook and Field data sheet for each sample.

8.0 Stream Flow Measurement

Stream flow volumes shall be measured during surface water sampling activities. To minimize sediment disturbance during sampling, the stream flow measurements should be conducted either downstream from the sampling point or after the completion of sample collection. RMC uses an electronic flow meter. The procedure for measuring stream flows is as follows:

1: Measure the width of the stream and divide the width into 0.5 foot increments.

2: At the midpoint of each 0.5 foot increment, record the total depth of the stream. The water velocity shall be measured at 0.6 of the total height of the water (e.g. if the water is one foot deep the velocity is measured at a depth of 0.4 foot from the surface or 0.6 feet from the streambed).

3: Turn the electronic stream meter gauge on. Set the meter to record the average velocity. Insert the stream flow gauge into the water at the midpoint of each segment with the arrow pointing in the direction of flow. Measure the velocity for approximately one minute and record the average.

4: Calculate the stream flow by calculating the area of each 0.5 foot wide segment by multiplying the width times depth. To obtain the flow volume for each 0.5 wide segment multiply the area of the segment by the average flow velocity for the segment. To obtain the total stream flow, add the total stream flow for each segment. An Excel spreadsheet is typically used for the calculations.

Calculations:

Segment flow volume = depth of 0.5 foot segment x width x flow velocity (feet/sec.) = cubic feet/ second
Total flow volume = sum of segment flow volumes.

9.0 Labeling

Each sample will be legibly labeled with a permanent marker containing the following information:

- Sample identification;
- Project number/name;
- Analyses requested;
- Preservatives (if required);
- Date/time collected; and
- Samplers initials.

10.0 Documentation

Field activities shall be recorded in a hard bound field notebook and field data sheet. Field notes shall include all pertinent information including but not limited to:

- Date and time samples were collected;
- Physical description of sample area;
- Identification of samples collected;
- Total number of samples collected;
- Total number of samples collected from each sample location;
- Physical description of samples;
- Preservatives used for samples;
- Sample container types;
- Filtered vs. Unfiltered samples;
- Analysis to be performed;
- Weather conditions;
- Hand sketches of subject area(s); and
- Description and date of any photograph(s) taken.

Sample handling and Chain of Custody documentation shall be in accordance with RMC SOP 5 found in this document.

11.0 Demobilization

After Decontamination, sample equipment will be stored in the appropriate, clean containers. Any equipment that suffers damage or excessive wear while conducting sampling will be labeled and reported to the equipment manager for the necessary maintenance, repair and/or replacement.

RMC SOP 5
STANDARD PROCEDURES FOR SAMPLE HANDLING, DOCUMENTATION, AND SHIPPING

1.0 Purpose

This section describes the handling and documentation procedures that will be used once soil and water samples are collected. The procedures will ensure that samples are handled properly and that appropriate documentation is completed.

2.0 Sample Handling

All samples will be promptly placed into a cooler to maintain a temperature of 4°C. Typically, samples selected for chemical analysis will be delivered at the end of each day to the analytical laboratory. If they are not submitted to the laboratory on the same day, they will be stored in a refrigerator in a locked storage room until they can be delivered to the laboratory.

3.0 Sample Identification and Labeling

Soil samples will be labeled in such a way as to identify the area and depth from which they were taken. Water samples will be labeled as to identify when and where they were collected from. Duplicate samples will always be labeled in the same manner such that the laboratory cannot tell they are duplicate (i.e., as a "blind duplicate"). Each sample container will be immediately labeled with the following information:

- Project name
- Project number
- Sample identification
- Date and time collected
- Analysis requested
- Filtered or unfiltered (water)
- Samplers initials
- Preservative used (water)

This information will also be recorded in the field logbook and on a Field data sheet.

4.0 Custody Seals

Custody seals shall be used to prevent tampering and to maintain sample integrity. A seal shall be placed across the top of sample jars or across the seals of plastic sample bags. The seal shall placed on the sample container directly preceding sampling and will be signed and dated by the sampler who collected the sampler.

5.0 Chain-of-Custody (COC)

COC documentation will begin in the field for each sample submitted to the laboratory and will also be maintained by laboratory personnel. Samples that are submitted to AEC will use the COC provided by AEC. COC forms will be requested from labs other than AEC prior to sample collection. A COC for each sampling event will be completed and will accompany each sample batch to the analytical laboratory. Sample custody means that all samples will remain in the possession or observation of the sampler at all times, or in a locked facility until delivery to the analytical laboratory. A sample COC form is provided in Appendix D. Copies of the COC forms shall be stored in a three ring binder for sample tracking.

6.0 Field Book

RMC field personnel will maintain a field logbook to record all field activities. The field logbook will be a weather-resistant bound field book. All data generated during the project and any accompanying comments will be entered directly into the logbook in indelible ink; any corrections will be made with single line-out deletions. At no time will any pages be removed from the field logbook.

Each day's field activities will be documented, including the following minimum information:

- Date of field activity;
- Time of field activity;
- RMC field personnel's initials;
- Project name;
- Project number;
- Date and time samples were collected;
- Physical description of sample area;
- Identification of samples collected;
- Total number of samples collected per sampling event;
- Total number of samples collected from each sample location;
- Physical description of samples;
- Preservatives used for samples;
- Sample container types;
- Filtered vs. Unfiltered samples (water);
- Analysis to be performed;
- Weather conditions;
- Hand sketches of subject area(s); and
- Description and date of any photograph(s) taken.

7.0 Field Data Sheets

Field data sheets will be used to collect and organize data in the field. Field data sheets will be completed for each sample location. Completed originals of the field data sheets shall be placed in a three ring binder and numbered chronologically in a three ring binder to be archived in RMC's office. A set of copies will be stored in a three ring binder for on-site reference.

8.0 RMC Sample Logbook

RMC will maintain a sample logbook, which will track all samples collected and/or accepted by RMC. The logbook will provide a unique, six digit alphanumeric identifier that will be assigned to each sample collected. All samples collected will be assigned an identifier number, regardless of that samples' submission to a laboratory. The next available chronological number in the sample logbook will determine the identifier, and this number will be cross-referenced with a sample description number, assigned in the field.

The RMC Sample logbook will be a covered, bound journal with non-removable pages. At no time will any pages be removed from the sample logbook.
All entries into the sample logbook will be made in indelible ink; and all corrections shall consist of initialed, line-out deletion. Data contained therein will include:

- Unique identifier number;
- Date;
- Project number;
- Sample description number;

- Sampler initials; and Lab acceptance initials.

9.0 Shipping and Packaging

Prior to hand delivering or shipping samples, care will be taken to ensure that each sample container will be protected from punctures or breakage. Packing materials will be used when necessary. The sample containers will be placed in a cooler filled with ice sufficient to maintain the temperature of the sample at 4°C. The ice will be placed within plastic bags surrounding each sample container. For transport to out of town laboratories, the outside of the cooler will be taped shut.

9.1 Local Sample Delivery

All procedures to protect the sample containers and maintain sample temperature as described above will be performed. If the sample is to be delivered to a local laboratory, efforts will be made to deliver the samples on the same day as it was collected. If the samples cannot be delivered on the same day as they were collected, the samples will be locked in a refrigerator for overnight storage and delivered the next day to ensure that all holding times are met.

9.2 Out of Town Sample Delivery

All samples collected that are to be analyzed by out of town laboratories will be shipped the same day as collected. All shipments to out of town laboratories will be overnight shipments and received by the laboratory the next day. All precautions to protect and maintain sample temperature as described above will be taken. A chain of custody previously provided by the laboratory will be filled out and shipped with the samples. The laboratory will be contacted upon shipment of the samples.

SOP 6
STANDARD PROCEDURES FOR DECONTAMINATION

1.0 Purpose

This SOP details the Decontamination protocols for sampling equipment. In order to reduce the risk of transferring materials from one sample site to another, and to assure that there is no cross-contamination of samples, the following procedures will be used.

2.0 Decontamination Equipment

- 5 gallon buckets – For washing and the collection of rinsate.
- Alconox - Soap
- Scrub brushes – For cleaning sampling equipment.
- Distilled water – For final equipment rinse.
- Culinary tap water – for equipment rinse.
- Garbage bags – for clean equipment storage.

3.0 Decontamination Procedures

RMC uses the following decontamination procedure for equipment:

1. Gross contaminant removal

This step involves scrubbing the equipment using an Alconox and water solution and a stiff scrub brush. The scrubbing will continue until all visible contaminants are removed from the equipment. This water will be changed as necessary. The Alconox and water solution is typically prepared and stored in a clean 5-gallon bucket.

2. Clean detergent wash

This step involves using a clean volume of Alconox and water solution. Equipment will be washed in this solution once all gross contaminants have been removed during Step 1. This solution will also be changed as necessary. The Alconox and water solution is typically prepared and stored in a clean 5-gallon bucket.

3. Clear water rinse

This step involves rinsing the equipment in clear, culinary tap water. This water will be changed as necessary to maintain its purity. The water solution is typically collected and stored in a clean 5-gallon bucket.

4. Distilled water rinse

Distilled water will be used as a final rinse for all Decontamination procedures. The water will be poured from a new container, or sprayed from a suitable container or the equipment will be submerged in a suitable container. Decontamination (equipment) blanks will be collected as required in the Sampling and Analysis Plan. The water solution is typically collected and stored in a clean 5-gallon bucket.

4.0 Decontamination fluid disposal

Decontamination fluids shall be disposed of on-site in the tailings impoundment area.

SOP #SRC-OGDEN-01

Porewater Sampling from a Micro Push Point or Mini Piezometer

TABLE OF CONTENTS

1.0 PURPOSE

The purpose of this Standard Operating Procedure (SOP) is to provide a standardized method for collection of porewater samples from micro Push Points or mini piezometers, to be used by employees of USEPA Region 8, or contractors and subcontractors supporting USEPA Region 8 projects and tasks. This SOP describes the equipment and operations used for sampling porewater in areas which will produce data that can be used to support risk evaluations. Deviations from the procedures outlined in this document must be approved by the USEPA Region 8 Remedial Project Manager, Regional Toxicologist or On-Scene Coordinator prior to initiation of the sampling activity.

2.0 RESPONSIBILITIES

The Field Project Leader (FPL) may be an USEPA employee or contractor who is responsible for overseeing the porewater sampling activities. The FPL is also responsible for checking all work performed and verifying that the work satisfies the specific tasks outlined by this SOP and the Project Plan. It is the responsibility of the FPL to communicate with the Field Personnel regarding specific collection objectives and anticipated situations that require any deviation from the Project Plan. It is also the responsibility of the FPL to communicate the need for any deviations from the Project Plan with the appropriate USEPA Region 8 personnel (Remedial Project Manager, Regional Toxicologist or On-Scene Coordinator).

Field personnel performing porewater sampling are responsible for adhering to the applicable tasks outlined in this procedure while collecting samples. The field personnel should have limited discretion with regard to collection procedures, but should exercise judgment regarding the exact location of sample collection, within the boundaries outlined by the FPL.

3.0 EQUIPMENT

- Micro Push Point Sampers - installed as described in #SRC-OGDEN-06.
- Mini Piezometer - installed as described in #SRC-OGDEN-06.
- Syringe Assemblies - 50 ml, 100% polyethylene/polypropylene syringes configured to taigon tubing (1/4" OD x 1/8 ID) with clamps (to secure tubing to sampling port) and a stopper. The syringe assemblies are used to withdraw porewater and transfer the sample into the sample collection container.
- Syringes - used to withdraw additional porewater from sampling location. Made of 100% polyethylene/polypropylene and 50 ml in volume.
- Peristaltic Pump - used to purge groundwater through the push point sampler for sampling.
- Collection Containers - as specified in the Project Plan.

- Meter Stick - used to measure water depth, and water level.

- Decontamination Adapter - a cylindrical adapter used to backflush cleaning solution through the Push Point Samplers (PP27 and PP14) for decontamination.

- Gloves - for personal protection and to prevent cross-contamination of samples. May be plastic or latex; should be disposable and powderless.

- Field Clothing and Personal Protective Equipment - as specified in the Health and Safety Plan.

- Sampling Flags - used for identifying porewater sampling locations.

- Field Notebook - a bound book used to record progress of sampling effort and record any problems and field observations during sampling.

- Three-ring Binder Book- to store necessary forms used to record and track samples collected at the site. Binders will contain the Field Data Sheet, Site Diagram, and sample labels. Example forms are provided in the Sample Documentation SOP.

- Permanent Marking Pen - used to identify sample containers and for documentation of field logbooks and data sheets.

- Cleaning Solution - used to decontaminate the sampler after use. Obtained from the micro Push Point Sampler manufacturer, MHE Products, Inc..

- Deionized Water - used to rinse cleaning solution from the Push Point Samplers (PP27 and/or PP14) during decontamination.

- Trash Bag - used to dispose of gloves and any other non-hazardous waste generated during sampling.

- Plastic Waste Bottle - used to dispose of excess porewater collected and decontamination waste.

4.0 POREWATER SAMPLE COLLECTION

Collection of porewater samples occurs as two phases. First, the flux (flow of groundwater to surface water) is determined at a sampling location or stream reach according to #SRC-OGDEN-06. A porewater sample is collected ONLY if it is first determined that there is positive flux at this location or within the sampling reach (groundwater is moving towards the surface water body).

4.1. Preparation for Sample Collection

Described with the protocol for measurement of flux described in #SRC-OGDEN-06 (Figure 1).

4.2. Collection of Groundwater Flux Measurements

Described with the protocol for measurement of flux described in #SRC-OGDEN-06 (Figure 2).

4.3. Collection of Porewater Samples for Analysis

Remove the Taigon tubing used for flux measurement (Section 4.2) from the micro Push Point sample port and discard in the trash bag. Attach a syringe or peristaltic pump to the Push Point sample port. Withdraw water at a low-flow sampling rate (50-200 ml/min) (Figure 3). Once non-turbid aliquots have been withdrawn, remove the peristaltic pump or the syringe used to purge the pore water and attach a "syringe assembly" (a pre-assembled 50 ml, 100% polyethylene/polypropylene syringe clamped to Taigon tubing) to the sample port. Be sure to affix a clamp at the tubing mouth to ensure a good seal at the sample port.

Using the syringe, withdraw adequate amounts of porewater and transfer the sample (Figure 4) into the sample collection container specified in the Project Plan. Affix one sample ID label to the sample container, and one to the Field Data Sheet.

Care should be taken to avoid tracking sediment and/or silt from one area to another. As samples are taken sequentially, care should also be taken not to contaminate an area yet to be sampled with the residue of the sample that is currently being taken. In general one should move in a single direction through the sampling area. If an area is known or suspected of having a higher concentration of contaminants, all other considerations being equal, it should be sampled last to prevent cross contamination.

If sampling equipment is to be re-used, follow the decontamination procedures outlined in Section 8.0 before collecting the next sample.

5.0 SAMPLE CONTAINERS AND LABELING

Following the procedures outlined in Section 4.0, porewater samples are collected directly placed into sample containers, and shipped to the participating laboratory. For each porewater sample, two sample identification labels are required. One label is placed on the Field Data Sheet and the other label is affixed to the sample container.

Sample labeling will occur as prescribed below:

1) Place a pre-printed label onto the sample container (See Project Plan).
2) Place a pre-printed label onto the Field Data Sheet.
3) This procedure will be repeated for each porewater sample collected using clean sample containers and unique sample ID numbers.

Do not allow samples to freeze; place all samples directly onto wet ice (4°C). Ship samples under chain-of-custody, protected with suitable resilient packing material to reduce shock, vibration, and disturbance.

6.0 SITE CLEAN-UP

If any rinse water used for sample decontamination is generated in the course of sample collection, it must be disposed of as specified in the Project Plan.

All marker flags (if reused) should be decontaminated by wiping off with towels and/or baby wipes before re-use.

Disposable PPE and other non-hazardous waste generated during sampling activities will be placed in a trash bag and taken to a waste receptacle at the field office to prevent disturbance by animals and dispersion by wind. These wastes will be disposed along with trash at a municipal landfill.

Porewater and decontamination rinsate waste generated during sampling activities will be placed in DOT-compliant drums in accordance with 40 CFR 265 Part I. All non-hazardous waste will be disposed of in municipal waste bins.

7.0 RECORD KEEPING AND QUALITY CONTROL

Each field crew carries a three-ring binder book that contains the porewater field data sheet, site diagram, and sample labels. In addition, a field notebook is maintained by each individual or team that is collecting samples, as described in the Project Plan. Each porewater sample location is recorded on the site diagram. Each sample has an ID number affixed to the sample container, and the duplicate label must be affixed to the data sheet. Deviations from this sampling plan are noted in the field notebook, as necessary.

For each location, the notebook information must include:

a. date
b. time
c. personnel
d. weather conditions
e. sample identification numbers that were used
f. descriptions of any deviations to the Project Plan and the reason for the deviation.

Samples taken from areas with visible staining or other indications of non-homogeneous conditions should are noted. Field personnel will collect the proper type and quantity of quality control samples as prescribed in the Project Plan.

8.0 DECONTAMINATION

Because decontamination procedures are time consuming, having a quantity of sampling tools sufficient to require decontamination at a maximum of once per day is recommended. All sampling equipment must be decontaminated prior to reuse as prescribed in the Project Plan.

Equipment decontamination will consist of the following 4 steps:

1.) Cleaning Solution
2.) Deionized water rinse
3.) Acetone rinse
4.) Deionized water rinse

Begin decontamination of the micro Push Point Sampler by thoroughly removing all sand, silt etc. from the guard rod and the exterior of the Push Point Sampler. Connect the cleaning adapter to a "garden sprayer" (with the spray nozzle removed) filled with cleaning solution. Gently insert the screened-zone of the Push Point Sampler into the cleaning adapter, making sure not to bend the screened-zone. Push approximately 300 ml of pressurized cleaning solution through the sampler into a waste receptacle. Gently push the guard rod all the way into the bore of the Push Point sampler to dislodge any bridged material. Re-rinse the Push Point sampler with cleaning solution. Follow this with a distilled water/and or methanol rinse. Rinse the guard rod with cleaning solution, followed with a distilled water rinse then and acetone rinse followed by a second distilled water rinse.

Reinsert the guard-rod into the push point sampler and the device is ready for re-use.

Note: before the guard-rod is reinserted into the Push Point Sampler, all small bends in both the guard-rod and in the Push Point Sampler should be removed. Use caution when straightening the screened-zone, it is somewhat delicate without the guard-rod inside it, and can be broken through repeated bending. It is sometimes helpful when straightening the screened zone to insert the guard rod or the cleaning rod to the area of the bend in the screened zone. Gently unbend the portion of the screened zone nearest the rod and carefully advance the rod to the next bend. After the rod has been fully inserted into the screened zone perform the final screened zone straightening fine-tuning until the guard rod slides freely through it.

9.0 GLOSSARY

Project Plan - A written document that spells out the detailed site-specific procedures to be followed by the FPL and the field personnel. In this case, the Project plan consists of the Phase 3 Sampling and Analysis Plan.

10.0 REFERENCES

Henry, M. 2000. "MHE Push Point Sampling Tools". Proceedings of the Ground-Water/Surface-Water Interactions Workshop. July 2000. http://www.gsiwebpage.net accessed July 2001.

MHE Products, 2001. Operators Manual and Applications Guide. MHE-PP27" and MHE-PP14". Version 1.06. February 3, 2001.

FIGURES

Figure 1

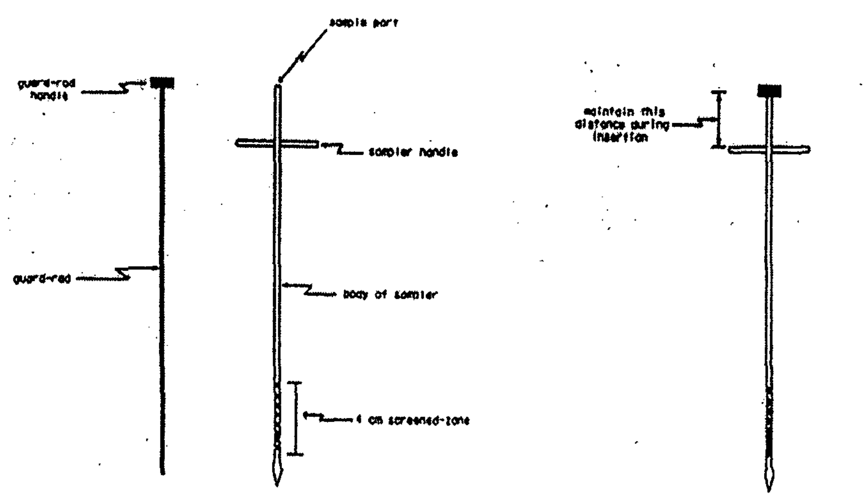


Fig. 1a
disassembled sampler

Fig. 1b
assembled sampler

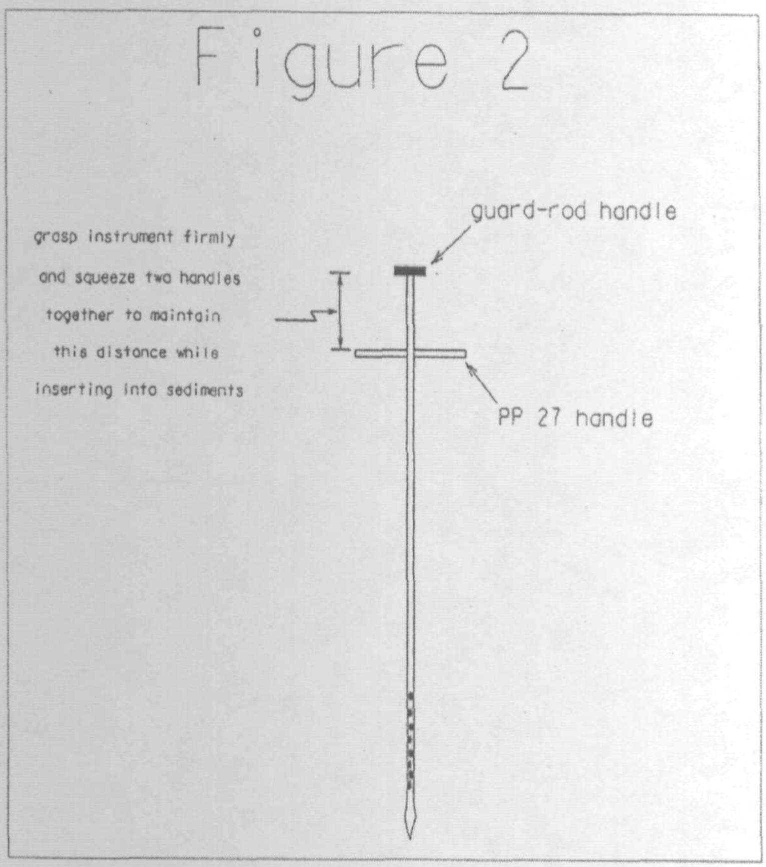
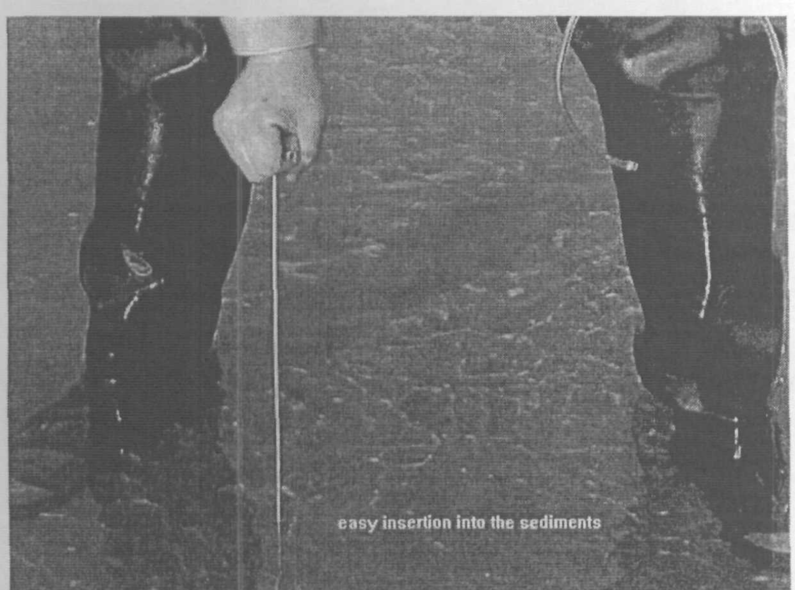


Figure 2
guard-rod handle
grasp instrument firmly
and squeeze two handles
together to maintain
this distance while
inserting into sediments
PP 27 handle


easy insertion into the sediments

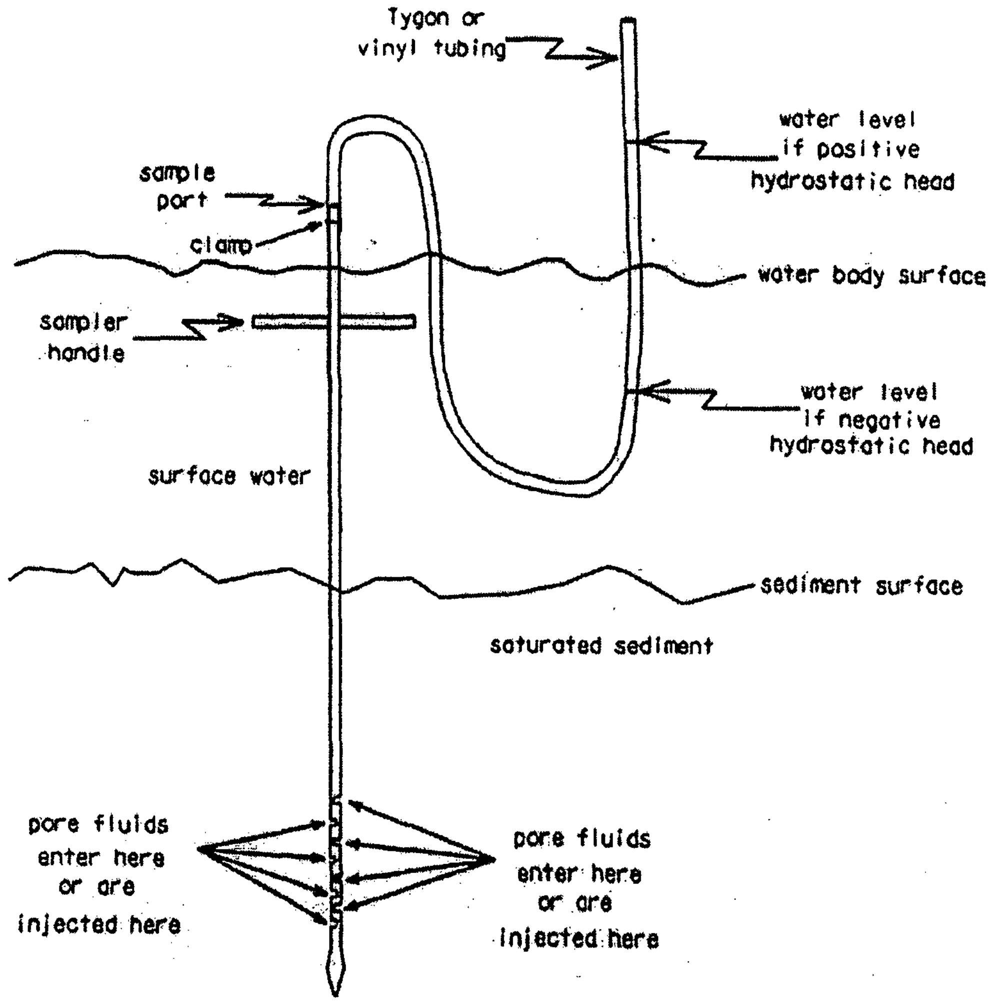

Tygon or
vinyl tubing
water level
if positive
hydrostatic head
sample
port
clamp
water body surface
sampler
handle
water level
if negative
hydrostatic head
surface water
sediment surface
saturated sediment
pore fluids
enter here
or are
injected here
pore fluids
enter here
or are
injected here

Figure 3

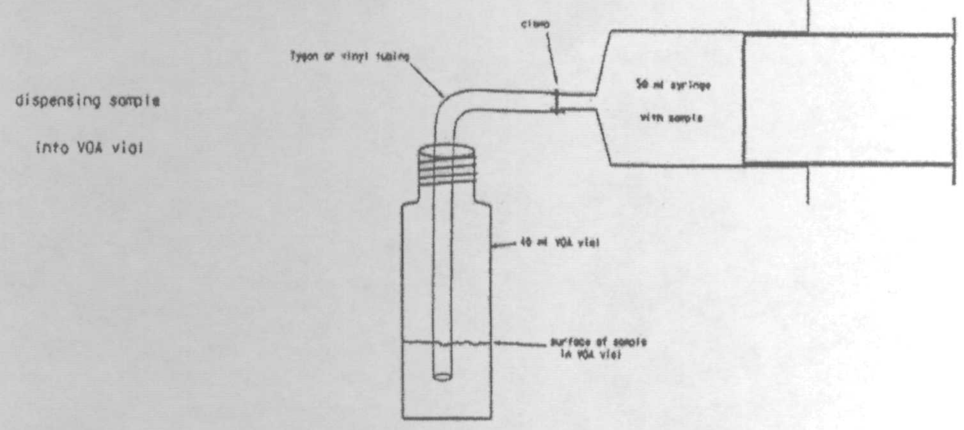
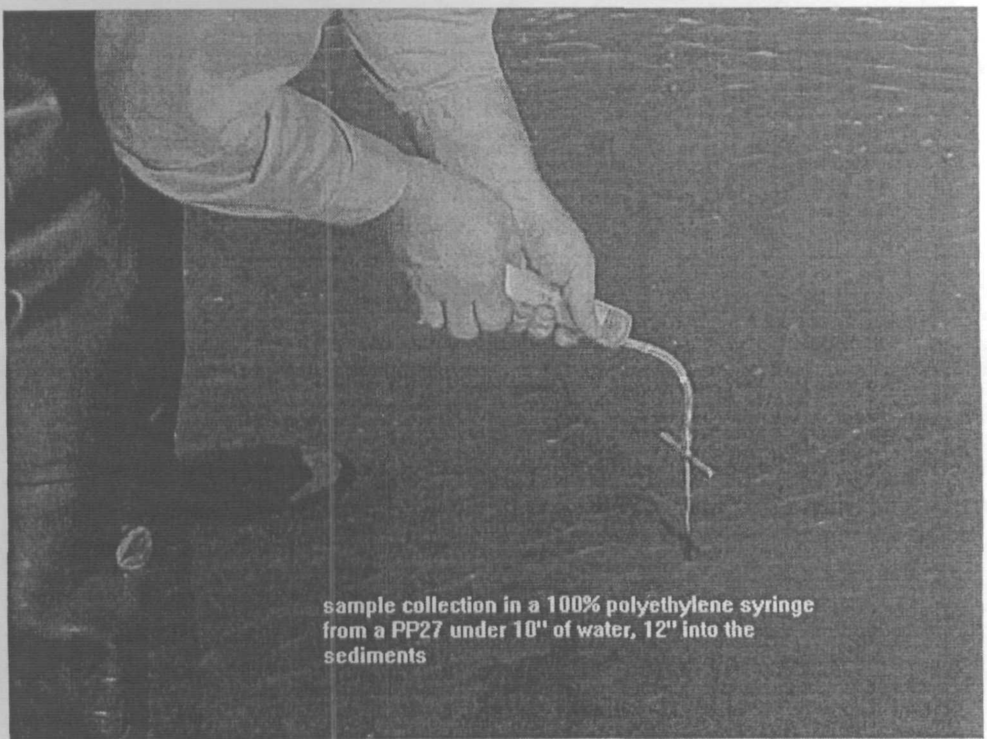

dispensing sample
into VOA vial
Tygon or vinyl tubing
clamp
50 ml syringe
with sample
40 ml VOA vial
surface of sample
in VOA vial

Figure 4


sample collection in a 100% polyethylene syringe
from a PP27 under 10" of water, 12" into the
sediments

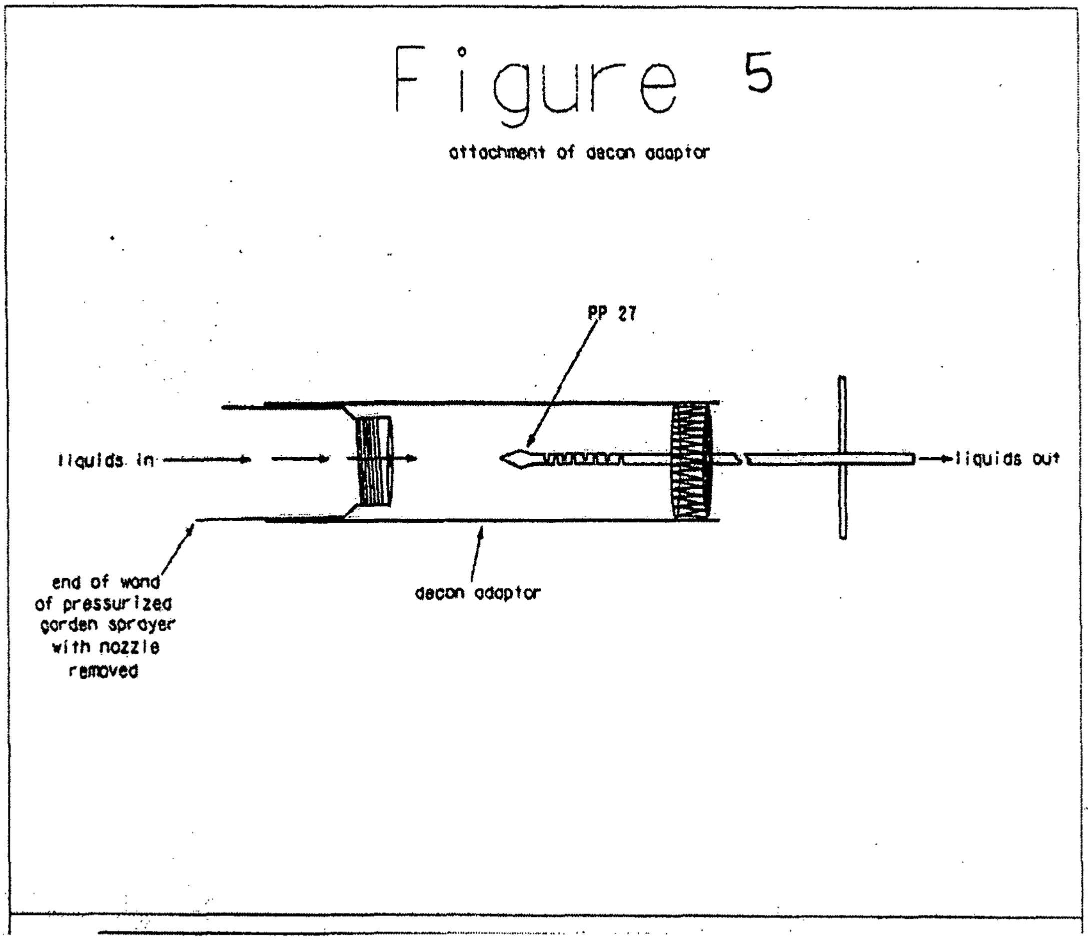


Figure 5
attachment of decon adaptor
PP 27
liquids in
liquids out
end of wand of pressurized garden sprayer with nozzle removed
decon adaptor

1.0 SCOPE AND APPLICATION

This standard operating procedure (SOP) describes the method for sampling terrestrial plant communities on hazardous waste sites. Analysis of vegetation will be used, in conjunction with other bioassessment techniques, to assess the impact of site contamination on plant life. Vegetation will be evaluated for shifts in community structure as a function of site contamination. Included below are procedures for obtaining representative measurements and guidance on quality assurance/quality control measures.

These are standard (i.e., typically applicable) operating procedures which may be varied or changed as required, dependent upon site conditions, equipment limitations, or limitations imposed by the procedure. In all instances, the ultimate procedures employed should be documented and associated with the final report.

Mention of trade names or commercial products does not constitute U.S. Environmental Protection Agency (U.S. EPA) endorsement or recommendation for use.

2.0 METHOD SUMMARY

The use of this SOP is dependent on weather and season. Non-woody plants will not endure throughout a winter with freezing temperatures, and thus cannot be evaluated by these methods in the winter.

A survey of site history will be made with all readily available information. Information on site contaminants, site and regional vegetation, and local climatic conditions will be considered. Remote sensing and topographic maps, when available, will be obtained and reviewed. Information on rare and endangered flora that may exist within the study areas should be obtained and reviewed.

Plots and transects are used to collect information representative of vegetative communities of the study site. Choice of appropriate sampling technique (i.e., plots vs. transects) depends upon site characteristics, plant characteristics, and study objectives. Information concerning species identification, enumeration, spatial arrangement, and size/shape attributes of the vegetation will be recorded in logbooks and on field data sheets. Signs of stressed vegetation will be noted. Samples representative of study location flora will be gathered for taxonomic verification. Values for species density, coverage, and frequency will be computed, as necessary.

3.0 SAMPLE PRESERVATION, CONTAINERS, HANDLING, AND STORAGE

Samples of vegetation may be required for taxonomic verification. Whole plants or selected parts (i.e., leaves, twigs, or flowers) will be placed in a resealable plastic bag and kept cool (4EC) to slow decay. All materials, with the exception of woody specimens, should be kept from temperature extremes and should be identified as soon as possible. If more than a week will pass before the samples can be identified, the samples will be placed in a plant press. Samples may also be archived by placing them in a plant press after identification.

4.0 INTERFERENCES AND POTENTIAL PROBLEMS

There are several potential problems and interferences that may occur when sampling plant communities.

1. Access to study locations must be obtained prior to study commencement.

2. Environmental disturbances, such as drought or fire, may confound data collection and interpretation. In addition, physical disturbances by man, such as the mowing or trampling of site vegetation, will further complicate assessment.

3. Microclimatic differences, such as sun/shade and moisture/drought, will affect plant growth and response.

5.0 EQUIPMENT/APPARATUS

Equipment needed for plant community sampling may include, depending upon the study objectives, the following items:

- Stakes - with sufficient height to be observed and sufficient width to stay in place during the period of study
- Line or rope
- Tape measure and/or plot frames
- Shovels and hand trowels - both of which must have unpainted stainless steel blades
- Pruning shears and/or knives
- Resealable plastic bags
- Cooler with ice
- Regional field guides to native plants
- Compass
- Vernier calipers
- Clinometer (optional) - necessary when measuring tree heights
- Documentation supplies (includes logbook, chain of custody records and custody seals, field data sheets and sample labels)
- Plant press (optional)

6.0 REAGENTS

Reagents are not required for preservation of vegetation samples. Samples should, however, be cooled to 4EC in order to minimize the degree of deterioration. Decontamination of sampling equipment may be required. Decontamination solutions are specified in ERT SOP #2006, Sampling Equipment Decontamination.

7.0 PROCEDURES

7.1 Sampling Considerations

7.1.1 General Site Survey

Prior to initiation of vegetation sampling, the appropriate sample collection area(s) should be determined. This may be accomplished with the assistance of remote sensing and/or topographic maps. Field guides to the regional vegetation species and experts knowledgeable about local conditions should be consulted. The extent of contamination should be established.

Consideration must also be given to the location of specific sampling points so that they provide representative samples (Section 7.1.2). The presence of rare or endangered species should also be determined and care taken not to adversely impact these communities during site activities.

A site sampling plan which details the number and general areas to be assessed will be prepared prior to plant community sampling activities.

7.1.2 Representative Samples

For representative sample collection, seasonal community fluctuations should be determined and climatic patterns analyzed. Topography and soil types should also be considered.

Sampling of vegetation should occur during seasons of the year where the species of interest are present. For example, if a complete vegetation survey were to be performed, plant assessment may be required over several seasons. If the species of concern were annuals, vegetation study should occur during the growing season while these species display characteristics that can be observed. Additionally, depending upon the study objectives, it may be necessary to survey plant communities several times during the growing season or throughout the year.

7.2 Sample Collection

The ecological parameters of density, coverage, and frequency reflect vegetational community structure and are those that are discussed in this SOP. Additional information may be collected for use in studies of plant community structure. Additional parameters useful in determining and comparing plant community structure include diversity and similarity indices. These parameters will not be addressed in the present SOP; however, measurements used to calculate these parameters may be collected at the same time as sampling activities described in this SOP. For a description of these additional parameters, refer to Brower and Zar.[1]

The size, shape, and number of vegetation sample locations ultimately depends upon the vegetation type present (i.e., herb, shrub, tree, vine, etc.) and their distribution pattern. Basically, there are two general approaches to plant community sampling: plots/quadrats and transects.

7.2.1 Sample Plots/Quadrats

A sample plot or quadrat is the specific area within which vegetation analysis will occur. The number, size, shape, and location of sample plots will depend upon the types of vegetation to be sampled and the objectives of the study. For example, smaller plots may be required for a site with dense or rich flora.

Typically, rectangular or circular plots are used. Circular plots are easy to set up. They require only a stake and premeasured line (or measuring tape). Circular plots are often used in the assessment of woody species. However, rectangular plots have been found, in general, to yield better results for plant surveys.(1) Rectangular plots require at least four stakes and a plot frame of desired size (or measuring tape and a means to make right angles) to be constructed.

The following procedure will be followed when surveying plant communities:

1. Divide vegetational areas of the site to be assessed into a grid. If soil/sediment sampling is also performed, it is most efficient and advantageous to use the same sample location grid for both soil/sediment sampling and plant community assessment. When vegetation is collected for analysis, use of the same grid locations will provide the potential for comparison of contaminant concentrations in the soil/sediment and the vegetation.

2. Select locations for a predetermined number of plots (as described in the site sampling plan) using randomly-selected grid coordinates. (X and Y coordinates can simply be paced out from the appropriate axis.)

3. Establish plots according to study objectives and the following vegetation classifications:

   a. <u>Closely Spaced Herbs</u> - [plants of less than 1 meter (m) in height]
      - use a rectangular plot of 1 m^2 (for example, 1.0 m x 1.0 m)

   b. <u>Bushes/Saplings/Shrubs</u> - [woody plants with height greater than 1 m and main stem diameter of less than 10 centimeters (cm), excluding vines]
      - use a plot area of 10 m^2 (for example, 2.5 m x 4.0 m)

   c. <u>Trees</u> - [any non-climbing woody plants with main stem diameter at breast height (DBH) of greater or equal to 10 cm. (DBH = 1.5 m above ground level)]
      - identify each tree within a 10 meter radius of the selected center point of the sample plot

   d. <u>Woody Vines (Lianas)</u> - (woody climber with DBH of less than 10 cm)
      - identify each vine within a 10 meter radius of the selected center point of the sample plot (usually associated with tree plots)

4. Identify and count species in each plot.

5. Estimate species coverage within plot area. Measure DBH for tree species, when applicable, to calculate basal area form which cover estimates are made.

6. Note visual cues of stress and overall health of plot vegetation (including wilting, browning, stunted growth, chlorosis, etc.).

7. Note habitat characteristics (for example, moisture availability, degree and direction of exposure of slope, tidal location, etc.).

8. Collect vegetative samples from each plot, as necessary, for taxonomic verification. Store samples as described in Section 3.0.

9. Repeat the above procedures for an uncontaminated reference area during the same period of study.

10. Perform appropriate calculations (Section 8.0) and appropriate statistical analyses upon the data.

11. Prepare generalized vegetation map showing

plant communities and sampling locations.

7.2.2 Transect Sampling

When the use of plots is impractical, transects may be used. Transects are especially useful in the evaluation of transitional communities. Ecological parameters that are studied utilizing plots can be studied utilizing transects. Additionally, changes in the vegetation in relation to environmental gradients may be observed. The type, size, number, and locations of transects chosen will depend upon study objectives, vegetation type, and site characteristics. Longer transects should be made when plants are widely dispersed.

Types of transects include belt transects and line intercept transects. A belt transect is a line transect with width. It is essentially a long, thin quadrat or can be divided into zones (each of which act as plots). In the line intercept method a known length of rope or tape measure is laid out in a line and information is collected as vegetation intercepts the line. The line intercept method is particularly useful for surveys of shrubs. This method is used for vegetative cover estimates and species composition estimates. With this method, only estimates of linear density can be made, as area is not involved.

The following procedure applies to plant community sampling using transects:

1. Determine which transect method best suits the objective(s) of the study and habitat available.

2. Establish transects according to the study objectives and the appropriate transect method:

   a. Belt transect

   - establish transect length and width
   - locate belt transect(s) randomly in the selected study area(s) or with bias along a specific gradient or feature of interest
   - identify and count species
   - estimate coverage and measure DBH (on woody species, when required) within plot(s)

   b. Line intercept

   - establish transect length
     Short lines (under 50 m) are used for assessment of herb species
     Long lines (greater then 50 m) are used for assessment of some shrub and tree communities
   - locate transect line(s) randomly in the selected study area(s) or with bias along a specific gradient or feature of interest
   - divide transect line into equal intervals
   - record the length of the line intercepted for each plant intercepting the line
   - count, measure, and identify plants that either intercept the transect line or are within a small distance from the line, depending upon the density of the vegetation

3. Note visual cues of stress and overall health of plot vegetation (including wilting, browning, stunted growth, chlorosis, etc.).

4. Note habitat characteristics (for example, moisture availability, degree and direction of exposure of slope, tidal location, etc.).

5. Collect vegetative samples from each transect, as necessary, for taxonomic verification. Store samples as described in Section 3.0.

6. Repeat the above procedures for an uncontaminated reference area during the same period of study.

7. Perform appropriate calculations (Section 8.0) and appropriate statistical analyses upon data.

8. Prepare a generalized vegetation map showing plant communities and sampling locations.

7.3 Sample Collection Variation

Taxonomic identification to the species level is often required for the vegetation assessment methods described. When no such knowledge is desired and/or available, a generalized physiognomic approach may be utilized. Physiognomy is the study of form, structure, and spatial arrangement of an organism.

The resulting data may be sufficiently detailed and organized and can be collected comparatively rapidly.

Physiognomic characteristics that may be observed and documented include:

- C Life form - presence, dominance, or absence of specific structural life forms (herbs, trees, vines, etc.)
- C Stratification and zonation - layers of vegetation from the ground-layer to the canopy
- C Foliage density - amount of shading vs. light penetration
- C Coverage - sparse (less than five percent coverage) to dense (greater than 75% coverage)
- C Dispersal pattern - arrangement of species (rows, clumps, solitary, etc.)
  - uniformity (evenly-spaced vs. irregularly distributed)
  - spacial separation (distant vs. dense)

8.0 CALCULATIONS

8.1 Calculations for Plots and Belt Transects

Density for Species i (D_i)

$D_i = n_i/A$ Where:
n_i = total individuals for species i
A = total area sampled

Relative Density for Species i (RD_i)

$RD_i = n_i/En$ Where:
n_i = number of individuals of species i
En = total number of individuals of all species in sampled plots

Coverage for Species i (C_i)

$C_i = a_i/A$ Where:
a_i = total area covered for species i
A = total area sampled

Relative Coverage of Species i (RC_i)

$RC_i = C_i/EC$ Where:
C_i = coverage for species i
EC = sum of coverage for all species

Frequency of Species i (f_i)

$f_i = j_i/k$ Where:
j_i = number of plots containing species i
k = total number of plots

Relative Frequency of Species i (RF_i)

$RF_i = f_i/Ef$ Where:
f_i = frequency of species i
Ef = sum of frequencies of all species

8.2 Calculations for Line Transects

Linear Density Index of Species i (ID_i)

$Id_i = n_i/L$ Where:
n_i = number of individual of species i
L = total length of all sampled transects

Relative Density for Species i (RD_i)

$RD_i = n_i/En$ Where:
n_i = number of individual of species i
En = total number individuals of all species in sampled transects

Linear Coverage Index of Species i (IC_i)

$IC_i = l_i/L$ Where:
l_i = sum of intercept lengths intercepted by species i
L = total length of all sampled transects

Relative Coverage of Species i (RC_i)

$RC_i = l_i/El$ Where:
l_i = sum of intercept lengths intercepted by species i
El = sum of intercept lengths for all species intercepting transects

Frequency of Species i (f_i)

$f_i = j_i/k$ Where:
j_i = number of intervals containing species i
k = total number of intervals on transects

Relative Frequency of Species i (RF_i)

$RF_i = f_i/\Sigma f$ Where:
f_i = frequency of species i
Σf = sum of frequencies of all species

8.3 Additional Calculation for Tree Species

Basal Area at Breast Height (A), calculated for each tree

$A = pi\ (r^2)$ Where:
pi = 3.1416
r = radius (in cm)

9.0 QUALITY ASSURANCE/ QUALITY CONTROL

The following quality assurance/quality control procedures apply:

1. All data must be documented on field data sheets or within field/site logbooks.

2. All instrumentation must be operated in accordance with the operating instructions as supplied by the manufacturer, unless otherwise specified in the work plan. Equipment checkout and calibration activities must occur prior to sampling/operation and they must be documented.

3. Calculations will be checked by an additional person at a rate of ten percent.

4. A sampling plan, including sample size, will be created prior to sampling.

10.0 DATA VALIDATION

Data generated will be reviewed according to the quality assurance/quality control considerations listed in Section 9.0.

In addition, taxonomic information will be confirmed by a regional biologist familiar with the site's vegetation.

11.0 HEALTH AND SAFETY

When working with potential hazardous materials, follow U.S. EPA, OSHA, and corporate health and safety procedures.

When sampling at a known or suspected contaminated site, precautions must be taken to safeguard the samplers from chemical and physical hazards. In addition, it would benefit the samplers to be familiar with and avoid any contact with plants that present a contact hazard such as poison ivy, poison sumac, and poison oak.

12.0 REFERENCES

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SOP BI-01
DECONTAMINATION OF TISSUE SAMPLING EQUIPMENT

Whenever possible, it is best to have the analytical laboratory process the tissue sample under clean laboratory conditions to limit possible contamination from field processing. If, however, the tissue sample must be processed in the field, then the following decontamination procedures should be followed (see project-specific field sampling plan [FSP] to determine if field processing is required).

To prevent potential cross contamination of samples, all reusable field filleting and tissue processing equipment (e.g., knives, tweezers, spoons) will be decontaminated before each use (i.e., before processing begins and between each sample). The cutting board will be decontaminated before processing begins and a piece of clean, unused aluminum foil will be wrapped on top of the cutting board (dull side up) for each sample (i.e., the foil will be replaced after each sample has been processed). At the sample collection site, a decontamination area will be established in a clean location, upwind of the actual sample processing location, if possible. This is where all field filleting and tissue processing equipment will be cleaned. Decontaminated equipment will be stored away from areas that may cause recontamination. Rinsate blanks will be collected according to SOP BI-02, *Preparation of Field Quality Control Samples—Tissue*. When handling decontamination chemicals, field personnel will follow all relevant procedures and will wear protective clothing as stipulated in the site-specific health and safety plan.

This SOP describes procedures for decontaminating field filleting and tissue processing equipment contaminated by either inorganic or organic materials. Sampling equipment used for both can combine these procedures, following the order of a detergent wash, site water rinse, organic solvent rinses, and final distilled/deionized water rinse with an acid rinse of all stainless-steel equipment at the end of each tissue processing field day.

EQUIPMENT REQUIRED

Equipment required for decontamination includes the following:

- Plastic tub (to collect solvent rinsate)
- Plastic bucket(s)
- Tap water or site water
- Carboy, distilled/deionized water (analyte-free; received from testing laboratory or other reliable source)

- Properly labeled spray bottles
- Funnels
- Alconox®, Liquinox® equivalent detergent
- Pesticide-grade ethanol and heptane (consult the project-specific FSP as the solvents may vary by EPA region or state)
- 10 percent (v/v) normal nitric acid (trace metal grade) for inorganic contaminants
- Baking soda
- Hard-bristle scrub brush
- Plastic sheeting, garbage bags, and aluminum foil
- Core liner caps or plastic wrap and rubber bands
- Personal protective equipment as specified in the health and safety plan.

DECONTAMINATION PROCEDURES FOR FULL SUITE ANALYSIS (ORGANIC AND INORGANIC ANALYTES)

The specific procedures for decontaminating field filleting and tissue processing equipment are as follows:

1. Rinse the field filleting and tissue processing equipment thoroughly with tap or site water to remove most of the remaining gross solids. Pieces that do not need to be used again that day may be set aside and thoroughly cleaned in the field laboratory at the end of the day.
2. Pour a small amount (i.e:, 0.5 teaspoon) of concentrated industrial detergent into a bucket and fill it with tap or site water. If the detergent is in crystal form, all crystals should be completely dissolved prior to use.
3. Scrub the equipment in the detergent solution using a brush with rigid bristles. Be sure to scrub with a back-and-forth motion.
4. Double rinse the equipment with tap or site water and set right-side-up on a stable surface to drain. Do not allow any surface that will come in contact with the tissue to touch any contaminated surface.
5. Carefully rinse the field filleting and tissue processing equipment with ethanol from a squirt bottle, and let the excess solvent drain into a waste container (which may need to be equipped with a funnel). Ethanol acts primarily as a drying agent, but it also works as a solvent for some organic

contamination. Set the equipment in a clean location and allow it to air dry. Use the ethanol sparingly and allow the ethanol to cascade down the surface of the equipment (i.e., the ethanol will "sheet" over the equipment surface).

6. Carefully rinse the air-dried field filleting and tissue processing equipment with heptane from a squirt bottle, and let the excess solvent drain into the waste container (which may need to be equipped with a funnel). The opening of the squirt bottle may need to be widened to allow enough solvent to run through the core liners without evaporating. Heptane acts as the primary organic solvent, but it is insoluble with water. If water beading occurs, it may mean that the equipment was not thoroughly rinsed with ethanol. When the equipment has been rinsed with heptane, set it in a clean location and allow the heptane to evaporate before using the equipment for tissue processing. Use the heptane sparingly and allow the heptane to cascade down the surface of the equipment (i.e., the heptane will "sheet" over the equipment surface).

7. Rinse the field filleting and tissue processing sampling collection with a final distilled/deionized water rinse.

8. If the decontaminated field filleting and tissue processing equipment is not to be used immediately, wrap them in aluminum foil (dull side facing the cleaned area).

9. If the field filleting and tissue processing equipment are cleaned at the field laboratory and transported to the site, then the decontaminated equipment will be wrapped in aluminum foil (dull side facing the cleaned area) and stored and transported in a clean plastic bag until ready for use, unless the project-specific FSP lists special handling procedures.

10. Rinse all stainless-steel equipment at the end of each sampling day with 10 percent (v/v) normal nitric acid solution.

After decontaminating all of the field filleting and tissue processing equipment, the disposable gloves and used foil will be placed in garbage bags and disposed of in a solid waste landfill. When not in use, keep the waste solvent container closed and store in a secure area. The waste should be transferred to empty solvent bottles and disposed of at a licensed facility per the procedures listed in the project-specific FSP. When not in use, keep the waste acid container closed and store in a secure area. The acid waste should be neutralized with baking soda and disposed of per the procedures listed in the project-specific FSP.

DECONTAMINATION PROCEDURES FOR INORGANIC ANALYTES ONLY

The specific procedures for decontaminating field filleting and tissue processing equipment are as follows:

1. Rinse the field filleting and tissue processing equipment thoroughly with tap or site water to remove most of the remaining (i.e., gross) solids. Pieces that do not need to be used again that day may be set aside and thoroughly cleaned in the field laboratory at the end of the day.
2. Pour a small amount (i.e., 0.5 teaspoon) of concentrated industrial detergent into a bucket and fill it with tap or site water. If the detergent is in crystal form, all crystals should be completely dissolved prior to use.
3. Scrub the equipment in the detergent solution using a brush with rigid bristles. Be sure to scrub with a back-and-forth motion.
4. Double rinse the equipment with tap or site water and set right-side-up on a stable surface to drain. Do not allow any surface that will come in contact with the tissue to touch any contaminated surface.
5. Carefully rinse stainless-steel field filleting and tissue processing equipment with a 10 percent (v/v) normal nitric acid solution from a squirt bottle, and let the excess acid drain into the waste container (which may need to be equipped with a funnel). Use the acid sparingly and allow the acid to cascade down the surface of the equipment (i.e., the acid will "sheet" over the equipment surface).
6. Rinse the field filleting and tissue processing equipment with a final distilled/deionized water rinse.
7. If the decontaminated field filleting and tissue processing equipment is not to be used immediately, wrap them in aluminum foil (dull side facing the cleaned area).
8. If the field filleting and tissue processing equipment are cleaned at the field laboratory and transported to the site, then the decontaminated equipment will be wrapped in aluminum foil (dull side facing the cleaned area) and stored and transported in a clean plastic bag until ready for use, unless the project-specific FSP lists special handling procedures.

After decontaminating all of the field filleting and tissue processing equipment, the disposable gloves and used foil will be placed in garbage bags and disposed of in a solid waste landfill. When not in use, keep the waste acid container closed and store in a secure area. The acid waste should be neutralized with baking soda and disposed of per the procedures listed in the project-specific FSP.

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SOP BI-04
FISH COLLECTION PROCEDURES USING AN ELECTROSHOCKER

This SOP discusses the sampling of fishes by use of an electrofishing device referred to as an "electroshocker." The procedures for processing fish captured by an electroshocker are described in SOP BI-08, *Fish Processing Procedures*. The personnel performing the fish collection will wear protective clothing as specified in the site-specific health and safety plan.

SAMPLE COLLECTION USING AN ELECTROSHOCKER

An electroshocker is an active fish collection device that sends an electric current through the water, temporarily stunning or directing the movements of fish. Stunned fish are collected by using a dip net. Because an electric current is generated during sampling, several precautions must be taken when using an electroshocker to avoid being electrocuted. Electroshocking should not be conducted without knowledge of the safety procedures described below. All equipment should be maintained and operated according to the manufacturer's instructions.

Basic procedures for using electroshockers are described below. One of four general electroshocker configurations can be used for fish collections. A backpack-mounted electroshocker is used in shallow streams where wading is safe. A pram shocker is used when wading in small and medium-sized shallow streams. The pram is a small barge-mounted electrofishing unit that allows one or more fish collectors to work simultaneously without the encumbrance of backpack-mounted units. A bankside shocker offers alternative sampling flexibility in that it can be stationed along an embankment and deployed throughout small or medium-size streams by the use of handheld electrodes with extended conductor cables. Pram and bankside shockers offer more power output than backpack shockers and, as such, potentially pose higher risks. A boat electroshocker is used along shorelines of deeper or open waters where wading is not possible or safe.

Safety Precautions

Electrofishing is hazardous work. The following safety precautions must be taken when using an electroshocker:

1. Never electrofish alone. The buddy system must always be enforced.
2. Ensure that all persons in the sampling crew wear proper sampling attire.

3. Ensure that all members of an electrofishing crew understand the system they are using and the risks involved. Before a field operation begins, new crew members should receive orientation on equipment and procedures. At least one member of the electrofishing crew must have CPR and first aid training.
4. Ensure that people, livestock, or pets are not in the water either upstream or downstream from the sampling site.
5. Do not use the electroshocker during an electrical storm or periods of heavy rainfall.
6. Limit the number of sampling crew to maximize safety through increased freedom of movement on deck or in the stream and to reduce confusion.
7. Make sure that the person-in-charge has ultimate control of the power source.
8. Never reach into the water with hands or feet for any reason while the electrosystem is operating.
9. Turn off the electroshocker immediately if a person falls into the water. All sampling crew must know how to turn off the electroshocker.
10. When electroshocking in streams, proceed upstream at a slow pace. Do not chase the fish.
11. With the exception of standard shoreline fish community surveys, do not shock constantly; it is preferable to shock for a few seconds, stop shocking while continuing to move, and then begin shocking again.

Equipment Required

Equipment required for collecting samples with an electroshocker consists of the following:

- Electroshocker unit (backpack, pram, bankside, boat)
- Hip boots or chest waders (if wading)
- Rubber gloves designed specifically for electroshocking
- Dip nets
- Buckets.

Operating the Electroshocker

The electroshocker will be used to collect fish samples as follows:

1. Mark off the stream segment to be sampled, if applicable to the needs of the study.
2. Set up the electroshocking equipment according to the manufacturer's instructions. Each electroshocker configuration has unique set-up procedures.
3. Have all members of the sampling crew put on appropriate attire (e.g., gloves, chest waders, etc.).
4. Designate one person as the operator of the electroshocker (i.e., the "shocker").
5. Adjust the voltage and ampere settings to the appropriate levels for the conductivity and velocity of the water that will be sampled and the size range of the target fish. This decision is deferred to the experienced operator.
6. If so equipped, adjust the setting for the electroshocker timer to zero before each electroshocking effort to document "on-time" electrofishing effort.
7. Have the crew members that will collect the shocked fish (i.e., the "dip-netters") stand by with dip nets.
8. If sampling a small stream, have all sampling crew members enter the water at the downstream end of the survey stream segment.
9. In small streams, have the crew face upstream while the "shocker" begins moving the anode through the water by extending it in an upstream direction and then pulling it away from fish cover or back in a downstream direction. At the same time, have the "dip-netters" position themselves slightly downstream on either side of the "shocker" to capture the shocked fish and transfer them to collection buckets.
10. Have the sampling crew proceed in an upstream direction while electrofishing available fish micro-habitats until the end of the sampling segment is reached or until a pre-determined sampling time is expended. If electroshocking from a boat, the dip netters will position themselves at the handrail on the bow, from which point they can safely net the stunned fish.
11. Where quantitative fish data is not required, sample distances and times may be limited only by the needs of the survey.
12. When the end of the sampling segment is reached, record the number of electroshocker seconds elapsed during sampling plus the number of fish collected during that period.

13. Process the fish according to study design specifications and the procedures described in SOP BI-08, *Fish Processing Procedures*.

14. If replicate stream or shoreline segments will be sampled, repeat Steps 1–13 for each replicate.

RECORD KEEPING

In addition to the items presented in SOP GEN-01, *Field Documentation*, the following information should always be recorded in the field logbook for each sampling segment:

- Type of electroshocker used (backpack, pram, bankside, or boat)
- Length of the segment of shoreline that was sampled
- Water depth along the sampling segment
- The number of electroshocker seconds elapsed during sampling, plus the number of fish collected during that period
- Presence of sheen on surface of the water
- Physical/habitat quality of each sampling segment (use EPA nationally standardized method; U.S. EPA 1999) if required by project-specific FSP
- Number of individual fish collected and their respective species, each fish's length and weight, and if there are any physical anomalies (e.g., eroded fins, missing eyes, scoliosis or other body or mouth deformities, or skin lesions). If species identification is not definitive, save one specimen in a sample jar with site water (add preservative if available) for later confirmation by a second fisheries biologist.

SAMPLE CUSTODY AND SAMPLE SHIPPING

Sample custody will be maintained in accordance with procedures outlined in SOP GEN-02, *Sample Custody*. All samples will be packaged and shipped with other samples in accordance with procedures outlined in SOP GEN-03, *Sample Packaging and Shipping*.

FIELD QUALITY CONTROL SAMPLES

Details on collection of field quality control samples (e.g., replicate stream or shoreline segments, replicate fish within a stream or shoreline segment) will be specified in the project-specific FSP. Details on specific field quality control samples can be found in SOP BI-02, *Preparation of Field Quality Control Samples—Tissue*. Not all of the field quality control samples discussed in this SOP may be required for a given project. The specific field quality control samples will be identified in the project-specific field sampling plan (FSP) and quality assurance project plan (QAPP).

FIELD MEASUREMENTS

Depending on the specific project objectives, it may be necessary to perform field measurements of the *in situ* environment. Possible field measurements include concentration of dissolved oxygen, salinity, or conductivity in the overlying water. Details on collection of field measurements can be found in SOP SW-13, *Field Measurements for Surface Water.* The specific field measurements, if any, will be identified in the project-specific FSP.

DOCUMENTATION OF STATION LOCATIONS

The location of each area or transect electroshocked will either be well documented in the field logbook (including map of each area or transect) or be determined using a differential global positioning system (DGPS), which is capable of providing latitude and longitude coordinates with an accuracy of approximately 2 m. The DGPS consists of two satellite receivers linked to each other by a VHF telemetry radio system. The receiver will be on the sampling vessel. Details on collection of field station coordinates can be found in SOP GEN-04, *Station Positioning Using the Trimble Pathfinder Pro XRS.* The specific kind of station location documentation used for a given project will be identified in the project-specific FSP.

REFERENCES AND OTHER SOURCES

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SOP BI-05
FISH COLLECTION PROCEDURES USING A SEINE NET

This SOP discusses the sampling of fishes by use of a seine net. The procedures for processing the fish captured with a seine are described in SOP BI-08, *Fish Processing Procedures*. The personnel performing the fish collection will wear protective clothing as specified in the site-specific health and safety plan.

SAMPLE COLLECTION USING A SEINE NET

A seine net is used as an active sampling device to capture fish along a segment of shallow shoreline by encircling them. Each encircling effort or sweep of shoreline with the net is referred to as a "haul." The number of hauls and number of fish collected in each haul can be documented to yield quantitative (i.e., catch-per-unit-effort) information as a standard method of reporting fisheries seine data. Sampling by seine net is generally most effective in areas with smooth substrate and few underwater obstructions. A seine net consists of a length of mesh fabric, usually made of nylon or polyester, vertically suspended between a float line on top and a weighted lead line at the bottom. Seine nets can be obtained from commercial net vendors in various dimensions and mesh sizes. Seine nets commonly used for fisheries work have mesh sizes that range from 1/16 in. to 4 in. Specific seine dimensions are selectively used by stream investigators depending on the needs of the fish survey, fish sizes, or life stages of the fish sought.

Each end of the net is fastened to a metal or wooden pole referred to as a braille. Seine nets can be constructed with an extended bag at the center that aids in the entrapment of fish during the seine haul.

Equipment Required

Equipment required for collecting samples with a seine net consists of the following:

- Seine net (see project-specific FSP for correct mesh size and dimension)
- Brailles
- Hip boots or chest waders
- Collection buckets or sample containers

- Boat (for difficult areas to access)
- Tape measure or hip chain.

Sampling Procedures

Collection of fishes by using the seine net will proceed as follows:

1. Mark off the segment of shoreline to be sampled.
2. Hold the inner end of the seine at the beginning of the shoreline sampling segment.
3. Carry the other end of the seine into the water perpendicular to the shore (a second person is needed to complete this task). When sampling areas are difficult or dangerous to wade in, or when a very long seine (e.g., for seining an ocean beach) is deployed, a boat can be used to manipulate the outer end of the seine. When using a boat, one person should hold the seine pole while a second person rows the boat. Alternatively, the shoreward end of the seine can be tethered to a fixed object on the shore while the boat maneuvers the outer end of the seine.
4. Extend the seine away from shore until it is fully extended or until the water becomes too deep to maneuver the outer end of the net. Ideally, the water depth to be sampled is no deeper than the mesh wall on the seine net. If an extra bag is sewn into the seine net, make sure the bag is extended out behind the seine.
5. With the first person pulling the inner seine pole from shore and the second person pulling the outer pole in the water, drag the seine parallel to the shoreline for the length of the sampling segment. Make sure the lead line drags along the substrate so that fish cannot escape under the net.
6. If necessary, a third person can follow behind the seine as it is being pulled to free the net from any snags that are encountered.
7. When the end of the sampling segment is reached, swing the outer end of the seine shoreward and continue moving (sweeping) the seine toward shore until both ends meet at the shoreline.
8. Pull the remainder of the seine toward shore, making sure that the lead line drags along the substrate. It is important to keep the seine net in motion during the entire process so as not to lose fish from the net. The net tends not to entangle fish, therefore, they can easily swim out of the net if forward motion is paused.
9. Check the net for fish after the entire seine is brought onto the shore.

10. Transfer the captured individuals to collection buckets.

11. Process the fish in accordance with study design specifications (see project-specific FSP) and SOP BI-08, *Fish Processing Procedures*.

12. If replicate shoreline segments are to be sampled, repeat Steps 1–10 for each replicate segment.

13. If a quantitative analysis of the fish community is being conducted (i.e., catch-per-unit-effort, total enumeration, or mark-recapture), it is recommended that the upper and lower boundaries of the stream segment be blocked by nets of the same mesh as the seine net. These nets should be strung across the channel, ensuring that the bottom of the net contacts the sediments so fish cannot move out of the stream segment being sampled.

RECORD KEEPING

In addition to the items presented in SOP GEN-01, *Field Documentation*, the following information should always be recorded in the field logbook for <u>each</u> sampling segment:

- Seine mesh size and dimension used
- Length of the segment of shoreline that was sampled
- Water depth along the sampling segment
- Presence of debris (e.g., roots, boulders) that may have impaired seining ability along the sampling segment
- Presence of sheen on surface of the water
- Physical/habitat quality of each sampling segment (use EPA nationally standardized method; U.S. EPA 1999) if required by project-specific FSP
- Number of individual fish collected and their respective species each fish's length and weight, and if there are any physical anomalies (e.g., eroded fins, missing eyes, scoliosis or other body or mouth deformities, or skin lesions). If species identification is not definitive, save one specimen in a sample jar with site water (add preservative if available) for later confirmation by a second fisheries biologist.

SAMPLE CUSTODY AND SAMPLE SHIPPING

Sample custody will be maintained in accordance with procedures outlined in SOP GEN-02, *Sample Custody*. All samples will be packaged and shipped with other samples in accordance with procedures outlined in SOP GEN-03, *Sample Packaging and Shipping*.

FIELD QUALITY CONTROL SAMPLES

Details on collection of field quality control samples (e.g., replicate shoreline segments, replicate fish within a shoreline segment) will be specified in the project-specific FSP. Details on specific field quality control samples can be found in SOP BI-02, *Preparation of Field Quality Control Samples—Tissue*. Not all of the field quality control samples discussed in this SOP may be required for a given project. The specific field quality control samples will be identified in the project-specific field sampling plan (FSP) and quality assurance project plan (QAPP).

FIELD MEASUREMENTS

Depending on the specific project objectives, it may be necessary to perform field measurements of the *in situ* environment. Possible field measurements include concentration of dissolved oxygen, salinity, or conductivity in the overlying water. Details on collection of field measurements can be found in SOP SW-13, *Field Measurements for Surface Water*. The specific field measurements, if any, will be identified in the project-specific FSP.

DOCUMENTATION OF STATION LOCATIONS

The location of each seine net placement will either be well documented in the field logbook (including map of each seine net placement) or be determined using a differential global positioning system (DGPS), which is capable of providing latitude and longitude coordinates with an accuracy of approximately 2 m. The DGPS consists of two satellite receivers linked to each other by a VHF telemetry radio system. The receiver will be on the sampling vessel. Details on collection of field station coordinates can be found in SOP GEN-04, *Station Positioning Using the Trimble Pathfinder Pro XRS*. The specific kind of station location documentation used for a given project will be identified in the project-specific FSP.

REFERENCES AND OTHER SOURCES

Hayes, M.L. 1983. Active fish capture methods. pp. 123–145. In: Fisheries Techniques. L.A. Nielsen and D.L. Johnson (eds). Publication of the American Fisheries Society, Bethesda, MD.

U.S. EPA. 1999. Rapid bioassessment protocols for use in streams and wadable rivers: periphyton, benthic macroinvertebrates and fish. Second Ed. EPA 841-B-99-002. Prepared by M.T. Barbour, J. Gerritsen, B.D. Snyder, and J.B. Stribling. U.S. Environmental Protection Agency, Office of Water, Washington, DC.

Note: SOPs GEN-01, -02, -03; and BI-01 cited within.

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SOP BI-08
FISH PROCESSING PROCEDURES

This SOP discusses the procedures for making biological measurements of individual fish and for resecting fillets from individual fish for analysis of chemical concentrations in edible muscle tissue.

All fish samples will be packaged and shipped in accordance with procedures outlined in SOP GEN-03, *Sample Packaging and Shipping.* Sample custody will be maintained in accordance with procedures outlined in SOP GEN-02, *Sample Custody.* Field activities will be recorded in accordance with procedures outlined in SOP GEN-01, *Field Documentation.*

BIOLOGICAL MEASUREMENTS/OBSERVATIONS

The biological measurements and observations commonly made of individual fish include length, weight, gender, reproductive condition, presence or absence of physical anomalies, parasites, or disease, and age determined by using scales or hard body parts.

Equipment Required

Equipment required for making biological measurements and resecting fish fillets consists of the following:

- Measuring board
- Analytical balance (and calibrated taring weight, if required)
- Stainless-steel filleting knife
- Skinning pliers (if needed for removing catfish skins)
- Blunt-point forceps
- Fish scale-remover ("scaler")
- Fillet board
- Microprojector (for aging, if required)
- Coin envelopes (for scale collection, if required)

- Aluminum foil (consult the project-specific field sampling plan [FSP] as the foil requirements may vary by EPA region or state)
- Ziploc® bags
- Collection buckets.

Length and Weight Measurements and Other Observations

Length and weight measurements should be made on unpreserved fish as soon as possible after collection. Preservation techniques such as freezing and fixation with formalin and ethanol can alter length and weight measurements relative to the values that would be found for unpreserved individuals immediately after capture. The procedure described below for measuring length addresses total length (i.e., the distance from the most anterior part of the fish to the tip of the longest caudal fin ray):

1. Examine each fish for signs of physical anomalies, disease, or external parasites. Examples of physical anomalies include eroded fins, missing eyes, scoliosis or other body or mouth deformities, and skin lesions. Examples of disease symptoms include hemorrhagic sores, skin fungi, or grossly undernourished body condition. Examples of external parasites include attached leeches or worms, or cysts embedded in the skin or fin membranes, or inside the gills. Detailed observations should be noted on appropriate data sheets for each fish examined. Note the location of the anomalies (i.e., caudal fin, left mandible).
2. Place each fish on the measuring board, with its head touching the wall of the board and its side resting along the ruler of the board. Do not squeeze the head of the fish against the wall of the board.
3. Push the caudal fin together, and record the measurement for the longest part of the fin to the specified accuracy (e.g., the nearest 1.0 mm).
4. Place the balance tray on the analytical balance, and press TARE. Wait for a reading of 0.0 g.
5. Place the fish in the balance tray.
6. Allow the weight reading to stabilize, and record the weight to the specified accuracy (e.g., 1.0 g).

Fish Filleting Procedures

Fish are commonly filleted to resect edible muscle tissue for analysis of chemical concentrations. The filleting process is the same one used by fishermen to remove edible muscle tissue from fish. The results of the chemical analyses are therefore directly related to the tissue that is frequently consumed by humans. Filleting should occur after length and weight measurements and other observations have been recorded for each fish, as follows:

1. Decontaminate all filleting equipment (filleting knife, scaler, fillet board) using the procedures provided in SOP BI-01, *Decontamination of Equipment—Tissue*.
2. Cover the cutting board with a piece of aluminum foil, dull side facing up.
3. Place each fish on its side on the fillet board.
4. Remove all scales from the caudal fin to the head. Do not remove the skin from fish that are commonly eaten with the skin attached to the fillet. For species that are commonly skinned before eating (e.g., catfish), remove the skin from the entire fish by cutting the skin around the head and peeling it off with pliers.
5. Make a cut along the ventral midline of the fish from the vent to the base of the jaw.
6. Make a diagonal cut from the base of the cranium, following just behind the gill to the ventral side just behind the pectoral fin.
7. Remove the flesh and rib cage from each side of the fish by cutting from the cranium along the spine and dorsal fin to the caudal fin. Leave the ribs attached to the main fillet. When removing the fillet, it is common to leave the fatty "belly" meat on the fish carcass. Consult the project-specific FSP and quality assurance project plan (QAPP) regarding inclusion of belly meat or rib bones with the fillet portions, because this procedural requirement may vary among agencies.
8. If necessary (see project-specific FSP), wrap the fillets in aluminum foil with the dull side facing the tissue.
9. Label the wrapped sample according to the instructions provided in the project-specific FSP.
10. Place the labeled, wrapped sample in a labeled Ziploc® bag, and preserve as indicated in the project-specific FSP and QAPP.

Determination of Gender and Reproductive State

Gender and reproductive state will be determined as follows:

1. After filleting each fish, examine the gonads, and determine whether they are ovaries or testes. Record the gender of the fish.

2. Identify the reproductive state of the gonads according to the following scale:

   - **Stage I**—Ovaries are wine-colored and shaped like torpedoes, and no eggs are visible; testes are small, flat, whitish in color, and cling closely to the spine.
   - **Stage II**—Ovaries resemble those in Stage I, except that small black (but color may vary) eggs are visible to the naked eye; testes are swollen and milky in appearance.
   - **Stage III**—Ovaries are somewhat swollen and yellowish in color; testes are large, lobed, and freely emit a milky liquid.
   - **Stage IV**—Ovaries are greatly swollen, their texture resembles tapioca, and the largest eggs are transparent and more than 1 mm in diameter; testes are slack and contain an abundance of connective tissue.
   - **Stage V**—Ovaries are slack and contain only a matrix and a few residual eggs.

Age Determination

The age of fish is commonly determined by counting the number of annual check marks (i.e., annuli) on hard structures such as scales, spines, otoliths, vertebrae, and opercular bones. The procedures described below are based on the use of scales for age determination. If otoliths, opercular bones, or vertebrae are required for age analysis, follow procedures specified in Nielson et al. (1983) or as otherwise indicated in the project-specific FSP.

1. Only personnel experienced in the process of fish-scale age determinations should be assigned to this task. At least one experienced peer should validate age determinations.
2. Before collecting scales for age determinations, remove mucous, dirt, and epidermis from the area by gently wiping the side of the fish in the direction of the tail with a blunt-edged knife.
3. Remove about 20 scales from the left side of each fish from areas suitable for the particular species being aged. Consult standardized methods manuals or experienced fisheries workers to obtain this information. Removal must be done carefully. Blunt forceps or a knife tip may be very useful for this task. Be careful not to break the margins of the scales or scratch the surfaces. Scales that are broken or irregularly shaped should be discarded.
4. Transfer fish scales to a labeled coin envelope for later age determination. For bullheads and catfishes, remove the dorsal spine for age determination instead of the scales. If otoliths, opercular bones, or vertebrae are required

for age analysis, follow procedures specified in Nielson et al. (1983) or as otherwise indicated in the project-specific work plan.

5. A scale sample number should be included on the coin envelope for each fish sampled. The sample number should cross reference vital data for each fish including information such as species, length, weight, sex, date, location, and project number.

6. Scales should be inspected and cleaned before mounting them for microscopic viewing. If mucus, skin pigments, or dirt is present on the scale, soak them in water for about two hours, and scrub off any remaining deposits with a small brush or piece of cloth after the soaking period. Retain the best 5 to 10 scales for mounting and viewing.

7. Mount the viewing scales between two microscope slides, making sure that the scales do not overlap.

8. Project the mounted scales with a microprojector (microfiche reader) and identify the scale(s) that have a complete set of rings emanating outward from their center. The microprojector should provide an enlarged image to about 50 times the natural size of the scale.

9. The number of annual rings (annuli) on each scale are counted. Each "true" annulus represents one year of growth. Care must be taken not to misinterpret "false" annuli, "split" annuli, checks, crowded annuli, or accessory rings. An important consideration for aging fish via scale marks is to understand the time of annulus formation which can vary with latitude, spawning, migration, and feeding habits of the sampled fish population as well as with environmental data and water temperature range.

10. Scale and age data are recorded on a Scale Analysis Summary Sheet (Attachment 115-1). The scale analyst must sign and date the sample control sheet.

REFERENCES AND OTHER SOURCES

Nielson, L.A., D.L. Johnson, and S.S Lampton. 1983. Fisheries techniques. American Fisheries Society, Bethesda, MD.

NYSDEC. Fish preparation procedures for contaminant analysis. New York State Department of Environmental Conservation, Albany, NY.

ATTACHMENT BI-08-1

Scale Analysis Summary Sheet

Exponent®

Scale Analysis Summary Sheet

Project Name:______________________

Project Number:____________________

Water Body:_______________________

Sample No.	Date Collected	Location	Species	Length (mm)	Weight (g)	Sex	Age	Remark

Analyst:______________________ Date:________

Witness:______________________ Date:________

SOP BI-11
AQUATIC INVERTEBRATE PROCESSING PROCEDURES

This SOP discusses the procedures for processing captured aquatic invertebrates.

EQUIPMENT REQUIRED FOR SAMPLE HANDLING AND PROCESSING

Equipment required for aquatic invertebrate sample handling and processing includes the following:

- Measuring board
- Scale and appropriate calibration weights
- 8-oz glass jars or aluminum foil
- Cooler with ice
- Adhesive sample labels
- Camera and film
- Field collection forms.

AQUATIC INVERTEBRATE LENGTH AND WEIGHT MEASUREMENTS

In addition to the items presented in SOP GEN-01, *Field Documentation*, the following information should always be recorded in the field logbook for <u>each</u> sampling location:

1. Place each aquatic invertebrates on a measuring board, and record its total length to the nearest millimeter from the tip of its rostrum to the end of the telsun (central tail section or uropod).
2. Place a balance tray on an analytical scale, and press TARE. Wait for a reading of 0.0 g.
3. Place the aquatic invertebrates in the balance tray.
4. Allow the weight reading to stabilize, and record the weight to the specified accuracy (e.g., 1.0 g).
5. Record measurements in the field logbook.

6. Aluminum foil may or not be required; however, if it is required, wrap aquatic invertebrates in aluminum foil with the dull side touching the aquatic invertebrates. Place aquatic invertebrates in resealable plastic bag (consult the project-specific FSP, as the sample packaging requirements may vary by EPA region or state).
7. Place adhesive label on the outside of this plastic bag.
8. Labeled plastic bags should be placed in a clean, resealable plastic outer bag and stored on dry ice or wet ice pending shipment to the laboratory for tissue analysis.
9. If required sample sizes are greater than the mass of individual organisms, the composition of any composite samples should be noted in the field logbook (number of organisms, species, if possible).
10. Sample preparation and analysis, problems encountered, and corrective action taken during sample collection, preparation, and delivery shall be recorded in the field logbook.

At no time should organisms that are found dead in sample collection devices or that are known to have been caught more than 24 hours before collection be retained for analysis. Checking sample collection devices on a daily basis is required.

SAMPLE CUSTODY AND SAMPLE SHIPPING

Sample custody will be maintained in accordance with procedures outlined in SOP GEN-02, *Sample Custody*. All samples will be packaged and shipped with other samples in accordance with procedures outlined in SOP GEN-03, *Sample Packaging and Shipping*.

FIELD QUALITY CONTROL SAMPLES

Details on collection of field quality control samples (e.g., replicate sample collection device placements, replicate aquatic invertebrates within a sample collection device) will be identified in the project-specific FSP. Details on specific field quality control samples can be found in SOP BI-02, *Preparation of Field Quality Control Samples—Tissue*. Not all of the field quality control samples discussed in this SOP may be required for a given project. The specific field quality control samples will be described in the project-specific FSP and quality assurance project plan.

FIELD MEASUREMENTS

Depending on the specific project objectives, it may be necessary to perform field measurements of the *in situ* environment. Possible field measurements include concentration of dissolved oxygen, salinity, or conductivity in the overlying water. Details on collection of field measurements can be found in SOP SW-13, *Field Measurements for Surface Water.* The specific field measurements, if any, will be identified in the project-specific FSP.

DOCUMENTATION OF STATION LOCATIONS

The location of each net or sample collection device placement will either be well documented in the field logbook (including map of each sample collection device placement) or be determined using a differential global positioning system (DGPS), which is capable of providing latitude and longitude coordinates with an accuracy of approximately 2 m. The DGPS consists of two satellite receivers linked to each other by a VHF telemetry radio system. The receiver will be on the sampling vessel. Details on collection of field station coordinates can be found in SOP GEN-04, *Station Positioning Using the Trimble Pathfinder Pro XRS.* The specific kind of station location documentation used for a given project will be identified in the project-specific FSP.

REFERENCES AND OTHER SOURCES

Hobbs, H.H., Jr. 1976. Aquatic invertebrateses (Astacidae) of North and Middle America. Water Pollution Control Research Series 18050 ELDO5/72. U.S. Environmental Protection Agency, Office of Research and Development, Cincinnati, OH.

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SOP BI-12
BENTHIC MACROINVERTEBRATE SAMPLING USING A GRAB SAMPLER

This standard operating procedure (SOP) describes the procedures used to sample benthic macroinvertebrate assemblages by using a grab sampler (e.g., modified van Veen, Ekman, Ponar). Benthic assemblages are typically analyzed for the abundances and biomass of various species and major taxa. The project-specific field sampling plan (FSP) should stipulate the number of replicate samples (i.e., individual grabs) that need to be collected at each station. The personnel performing the benthic macroinvertebrate collection and sample processing will wear protective clothing as specified in the site-specific health and safety plan.

All benthic macroinvertebrate samples will be packaged and shipped in accordance with procedures outlined in SOP GEN-03, *Sample Packaging and Shipping* with consideration of information provided in SOP HS-01, *Restricted Article Shipment*. Sample custody will be maintained in accordance with procedures outlined in SOP GEN-02, *Sample Custody*. Field activities will be recorded in accordance with procedures outlined in SOP GEN-01, *Field Documentation*.

The grab sampler used for benthic infauna studies should be capable of collecting acceptable samples from a variety of substrates, including mud, sand, gravel, and pebbles (APHA 1989). The procedures for sampling benthic macroinvertebrate assemblages by using a grab sampler are described below.

EQUIPMENT REQUIRED

Equipment required for benthic macroinvertebrate sampling includes the following:

- Grab sampler (e.g., modified van Veen, Ekman, Ponar)
- If grab sampler is of considerable weight, then a winch and hydrowire (with load capacities ≥3 times the weight of a full sampler)
- Sample collection table (if vessel deck space allows)
- Sample collection tub
- Ruler
- Sieve(s) (typically with a 0.595-mm mesh for freshwater studies or a 1.0-mm mesh for marine studies; consult project-specific FSP for correct sieve size);

multiple sieves can be stacked on top of each other to capture different size fractions of benthic macroinvertebrates that will be processed separately; consult project-specific FSP for correct number of sieves

- Scoop (for transferring sediment sample aliquots to the sieve)
- Sample containers (clean, 1-L wide mouth plastic jars with plastic screw-on lids)
- Internal labels
- 10 percent buffered formalin
- Rose bengal (depending on study objectives, rose bengal stain may or may not be added; consult project-specific FSP)
- Scrub brush and soft-bristle nylon brush or tooth brush
- If necessary, socket and crescent wrenches (for adding or removing the detachable weights of the van Veen grab sampler)
- Water pump and hose (for sieving samples and for rinsing the grab sampler, sample collection tub, and sample collection table).

GRAB SAMPLER DEPLOYMENT

1. Prior to deployment, clean the inside of the grab sampler with a scrub brush and site water.
2. Depending on the sampling environment and substrate, consult either SOP SD-04 *Surface Sediment Sampling Using a Modified van Veen Grab Sampler*, SOP SD-05, *Surface Sediment Sampling Using an Ekman Grab Sampler*, or SOP SD-06, *Surface Sediment Sampling Using a Ponar Grab Sampler* for the correct deployment techniques for the appropriate grab sampler.
3. Lower the sampler through the water column at a slow and steady speed (e.g., 30 cm/second).

Allow the grab sampler to contact the bottom gently, with only its weight being used to force it into the sediments. The sampler should never be allowed to "free fall" to the bottom because this may result in premature triggering, or improper orientation upon contact with the bottom.

GRAB RETRIEVAL

1. After the grab sampler has rested on the bottom for approximately 5 seconds, begin retrieving it at a slow and steady rate (e.g., 30 cm/second).
2. Ensure that the sampling vessel is not headed into any waves before the sampler breaks the water surface to minimize vessel rolling and potential sample disturbance.
3. After the grab sampler breaks the water surface and is raised to the height of the sample collection table or sample collection tub, rinse away any sediments adhering to the outside of the grab sampler (it is essential that the sediments adhering to the outside of the grab are removed because those sediments and any associated benthic macroinvertebrates are not part of the sample).
4. After rinsing is finished, raise the grab sampler above the height of the collection table or sample collection tub, swing it inboard, and gently lower it into the sample collection tub on the sample collection table while maintaining tension on the hydrowire to prevent the grab sampler from rolling when it contacts the bottom of the tub.
5. When the grab sampler contacts the bottom of the table or tub, insert wedges under both jaws, if necessary, so that the grab sampler will be held in an upright position.
6. Open the doors on the top of the grab sampler, and inspect the sample for acceptability. The following acceptability criteria should be satisfied:
   - The sampler is not overfilled with sample to the point that the sediment surface presses against the top of the sampler or is extruded through the top of the sampler (organisms may have been lost)
   - Overlying water is present (indicating minimal leakage)
   - The overlying water is not excessively turbid (indicating minimal disturbance or winnowing)
   - The sediment surface is relatively undisturbed; the sediment-water interface is intact and relatively flat with no sign of channelling or sample washout
   - The desired penetration depth is achieved (see project-specific FSP); the following penetration depths should be achieved at a minimum:
   - 4–5 cm for medium-coarse sand
   - 6–7 cm for fine sand
   - >10 cm for silty sediment

- There is no sign of sediment loss (incomplete closure of the sampler, penetration at an angle, or tilting upon retrieval).

If a sample fails to meet the above criteria, it will be rejected and discarded away from the station. The location of consecutive attempts should be as close to the original attempt as possible, and if sampling on a river or stream, consecutive attempts should be located in the "upstream" direction of any existing current. Rejected sediment samples should be discarded in a manner that will not affect subsequent samples at that station or other possible sampling stations.

Penetration depth should be determined by placing a ruler against the center of the inside edge of the opening on the top of one side of the grab sampler and extending it into the grab sampler until it contacts the top of the sample. The penetration depth is determined by the difference between that measurement and the total depth of the grab sampler.

SAMPLE REMOVAL AND PROCESSING

1. For each acceptable sample, characterize the sample as specified in the study design. Characteristics that are often recorded include the following:

   - Sediment type (e.g., silt, sand)
   - Texture (e.g., fine-grain, coarse, poorly sorted sand)
   - Color
   - Biological structures (e.g., chironomids, tubes, macrophytes)
   - Approximate percentage of biological structures
   - Presence of debris (e.g., twigs, leaves, wood chips, wood fibers, manmade debris)
   - Approximate percentage of organic debris
   - Presence of shells
   - Approximate percentage of shells
   - Presence of a sheen
   - Odor (e.g., hydrogen sulfide, oil, creosote)
   - Changes is sediment characteristics
   - Presence and depth of redox potential discontinuity layer (if visible)
   - Maximum penetration depth

- Comments relative to sample quality (i.e., leakage, winnowing, disturbance).

2. After the sample is characterized, open the jaws of the grab sampler so that its contents (i.e., sediments <u>and</u> overlying water) are released into the sample collection tub.

3. Rinse any remaining sediment inside the grab into the collection tub, being careful not to overfill the tub with water.

4. Before each sample is sieved, all sieves will be examined for damage and wear. Look for rips in the mesh, irregular mesh spacing, and sand grains caught in the mesh. Use water pressure or a soft nylon brush to dislodge sand. DO NOT use sharp objects or stiff brushes, as the mesh may be damaged or torn.

5. After the entire sample has been collected in the sample collection tub, carefully transfer aliquots of the sample to the sieve by using a scoop.

6. Sieve each sample aliquot by rotating the sieve (in an up-and-down, not swirling, motion) in a bucket of water or by passing a gentle stream of water through the sieve from above or washed using a combination of these techniques. For all methods, it is imperative that the samples be washed gently to minimize specimen damage.

7. After each aliquot has been sieved, carefully rinse all of the retained material into a sample container, and carefully check the sieve to ensure that no organisms are trapped in its mesh (do not fill any sample container more than three-quarters full to ensure that a sufficient amount of space is available for the fixative).

8. If an organism is found to be trapped in the sieve, dislodge it with a gentle stream of water or by using forceps, and transfer it to the sample container.

9. Continue sieving aliquots of the sample until all of the sample has been processed.

10. Any large stones or other debris in the sample too large to fit in the sample jar should be thoroughly and carefully rinsed-off into the sieve, removed, and discarded under the supervision of the field team leader and noted on the field logbook.

11. After the entire sample has been sieved, clean the sieve by turning it over and back-washing it with a high-pressure spray to dislodge any sediment grains or detritus that are lodged in the mesh.

12. Fix each sample by filling each sample container with a 10–15 percent solution of borax-buffered formalin and inverting the container at least five times to ensure that the fixative penetrates all parts of the sample.

13. Depending on the sampling environment and the preferences of the taxonomic laboratory, the samples may be dyed with rose bengal (see project-specific FSP). If required, rose bengal should be added to the formalin solution prior to fixing the samples.

14. Label each sample container (both internal and external labels are required; see below), and store it in a protective container.

INTERNAL LABELS

In addition to the label on the outside of the sample container (i.e., external label, see SOP GEN-01, *Field Documentation*), a complete label must be placed inside each sample container. The internal label must be preprinted and should be made of at least 100 percent waterproof rag paper. The internal labels should be filled out using a pencil (i.e., no ink).

SAMPLE CONTAINERS

Samples can be stored in various containers including glass or plastic jars, and plastic bags. Exponent prefers that plastic jars with plastic screw-on lids (formalin corrodes metal) be used to store benthic macroinvertebrates samples. The use of this kind of sampling container lessens the possibility of formalin leakage during shipping and the breaking or tearing of the sample container. In general, a single 1- or 2-qt container is large enough to hold a sieved sample from a van Veen grab sampler and 1-L container is large enough to hold a sieved sample from a Ekman or Ponar grab sampler. If the sample volume exceeds one half of the container volume, more than one container should be used. Use of multiple containers for single replicates should be recorded in the field logbook.

After the buffered formalin has been added to a sample container, it is critical that the contents be mixed adequately. This usually can be accomplished by inverting the container several times (make sure that the lid is tightly screwed on). After mixing, sample container should be placed in protective containers for storage and transport o the laboratory. After being stored for approximately 1 hour, samples should be inverted several times again to ensure adequate mixing. Onboard the sampling vessel, samples should be stored so as to minimize exposure to sunlight and temperature extremes. They should also be stored in a stable part of the ship to minimize agitation.

BUFFERED FORMALIN PREPARATION

The fixative most commonly used for benthic macroinvertebrate samples is formalin, an aqueous solution of formaldehyde gas. Under no circumstances should ethyl or isopropyl alcohol be used as a preservative in place of the formalin. Penetration of the alcohol into body tissues is too slow to prevent decomposition of the specimens.

Solutions of 10–15 percent buffered formalin are most commonly used. However, samples containing large amounts of organic debris (e.g., peat, woody plant material) may require higher concentrations. The volume of fixative should be at least twice the volume occupied by the sample. If possible, the formalin solution should be added to the sample container until it is completely filled. This will minimize abrasion during shipping and handling. It is recommended that at least 2 L of diluted formalin solution be on hand for each replicate van Veen grab collected and at least 0.75 L of diluted formalin solution be on hand for each replicate Ekman or Ponar grab collected. The formalin solution should always be buffered to reduce acidity. Failure to buffer may result in decalcification of molluscs and echinoderms. Ideally, pH should be at least 8.2, as calcium carbonate dissolves in more acidic solutions. Borax (sodium borate) should be used as the buffer because other buffering agents may hinder identification by leaving a precipitate on body tissues.

To prepare a 10-percent buffered formalin solution, add 4 oz of borax to each gallon of concentrated formalin (i.e., a 40-percent solution of formaldehyde in water). This amount will be in excess, so use the clear supernatant when making seawater dilutions. Dilute the concentrate to a ratio of one part concentrated formalin to nine parts site water (sea water or tap water). If seawater is used, it will further buffer the solution. Fresh buffered formalin should be made prior to each sampling event, because formalin will eventually consume all of the buffering capacity of the borax.

If staining is used (see project-specific FSP), rose bengal is often added to the buffered formalin to facilitate sorting by staining the benthic organisms. The stain colors most infauna and thereby enhances their contrast with the debris from which they are sorted. Taxa that do not always stain adequately include ostracods and gastropods. **BE CAREFUL** when adding rose bengal to the buffered formalin solution. Add only a **VERY SMALL AMOUNT** of rose bengal; a little rose bengal goes a very long way. **REMEMBER**, you can always add more stain to the buffered formalin if you need to, but you can not remove the rose bengal once it has been added.

SOP BI-13
VEGETATION SAMPLING

This SOP describes the methodology for vegetation sampling and outlines procedures for determining the following vegetation measurements:

- Species richness
- Cover
- Frequency
- Biomass production
- Plant tissue analysis.

As used in this procedure, the term vegetation refers to terrestrial vascular plants unless otherwise specified. The methods in this SOP apply primarily to herbaceous plant communities and are not specifically directed at woody plant communities. Study design and data analysis are project specific and are not addressed in this SOP, but may be found in the project sampling and analysis plan (SAP). For each of the methods outlined below, random location of the samples is desired, and details related to locating samples will be provided in the SAP with the study design. Some modification of these procedures may be required depending on health and safety issues and the specific objectives outlined in the SAP.

For all vegetation fieldwork, voucher specimens should be made of all plant species not readily identifiable or known in the field. Voucher specimens should have the appropriate structures needed for laboratory identification (i.e., roots, stems, leaves, flowers, flower color, fruits, etc.). Specimens should be placed within individual labeled, folded sheets of newspaper and pressed by using proper botanical technique. Specific plant information on each collected voucher specimen should be recorded in the field notebook or the collector's plant collection notebook.

SPECIES RICHNESS DATA COLLECTION

Species richness can be determined through the use of relevé surveys, belt transects, point-intercept transects, or quadrats (plots). The specific method to be used on a project will be specified in the SAP.

Relevé Surveys

The relevé survey is conducted by compiling a comprehensive species list for each community or area of interest as specified in the SAP. The area to be surveyed is delineated and thoroughly walked through. During the walkthrough, all plant species observed in the area are identified and recorded on the datasheet. Abundance categories (Braun-Blanquet cover-abundance scale; Bonham 1989) may be assigned to each species if desired.

Belt Transects

The belt transect survey is conducted along a transect line. A transect line is laid out with a tape measure following the specific guidelines for length and transect location provided in the SAP and/or based on site-specific restrictions. Transect endpoints should be permanently marked and labeled to aid in relocation. Using the tape as a center line, identify and record on the data sheets all plant species rooted within one meter on either side of the tape (for the entire length of the transect). A 2-m long rod with the center point marked can be used to determine whether species near the edge are located within the 1-m outer boundary.

Point-Intercept Transects

The method for this type of transect is detailed in the section on cover. However, species richness is determined from the list of species for which foliar vegetation hits are recorded. The use of this method alone for species richness determination is not generally recommended, however, because the number of species encountered will be fewer than would be found by using the other methods listed here, and many of the less frequent occurring species will be missed.

Quadrats (Plots)

Quadrat or plot sampling for species richness consists of compiling a list of species found within a delineated area of specified size. Quadrats are generally square or rectangular in shape, although circular plots are also used. Common sizes for rangeland or herbaceous vegetation vary from 0.25 to 2 m^2. Quadrat size, the number of quadrats to sample, and the method for randomly locating quadrats for a given project are specified in the SAP. Species richness is determined by identifying and recording on the datasheet all plant species rooted within the quadrat.

COVER DATA COLLECTION

Cover is determined through the use of an optical point-intercept method or with the use of quadrats. The specific method to be used on a project will be specified in the SAP.

Optical Point-Intercept

The optical point-intercept method consists of evaluating what is encountered along a transect through the cross-hairs of an optical siting device. A transect is laid out with a tape, and sampling points are established at 0.5-m intervals along the transect line. Transect endpoints should be permanently marked and labeled to aid in relocation. The tripod with the optical siting device is positioned over the sample point on the transect line, and the optical device is positioned to the right of the transect. Observations are recorded on the datasheet as "hits" for each of the different cover categories, which include bare soil, litter, rock, lichen, moss, or vegetation. Hits on vegetation are recorded on the basis of species. Individual hits are considered to be indicative of the amount of cover provided by each of the cover categories. For example, if 100 points are evaluated along a transect, each hit is equivalent to 1 percent of the cover along the transect (1/100 of the total number of possible hits, or 1 percent). This method has an advantage over quadrat sampling in that it is more repeatable and consistent among different observers. The only decisions that need to be made are related to what appears in the cross-hairs of the siting device.

The number of transects to be sampled at each sampling area and the length of the transects are specified in the SAP. In some cases, however, the number and length of transects may depend on the size of available sampling area. Transect length can be adjusted to accommodate sample area size. However, as a general rule, 500 or more hits are recommended as a minimum to characterize an area. The number of transects and/or the linear distance between hits along the transect may be adjusted to properly characterize an area.

Quadrats (Plots)

See the discussion under *Species Richness Data Collection for Quadrats* for information on determining the size of and how to locate quadrats. Quadrat or plot sampling for cover consists of compiling a list of species found within a delineated area of specified size (i.e., a quadrat) and estimating the amount of coverage for each species. This information is recorded on the datasheet. Cover estimates are made for each species in the quadrat by using either the Braun-Blanquet cover-abundance scale (Bonham 1989) or the Daubenmire cover scale (Bonham 1989). The quadrat size, number of quadrats to sample, how to randomly locate the quadrats, and the abundance scale to use on a given project will be specified in the SAP.

This method provides a coarse estimate of cover and is not as repeatable as the optical point-intercept method. The first choice for estimating vegetation cover should be the optical point-intercept, but this method is provided for special situations in which that method is not desirable or feasible. As a general rule a minimum of 10 quadrats, but preferably 25–30 quadrats, is the best number for characterizing an area.

FREQUENCY

Species frequency information is obtained primarily from species richness or cover collection methods. Frequency provides an estimate of how often a species is encountered in an area given a specific quadrat size. Frequency is defined as how many times a species is encountered out of the total number of possible observations made (i.e., 20 quadrats out of 30 sampled). Often the final value is converted to a percentage (67 percent in this case). Values will vary depending on the actual method used to gather the species information (i.e., quadrats vs. point-intercept vs. belt transect). Therefore, using a consistent method when collecting data for frequency measurements is important for making valid comparisons. Quadrats can be added to a sampling protocol if specific frequency information is required and it cannot be adequately obtained from other data being collected. See quadrat sampling in the *Species Richness* and *Cover Data Collection* sections for more information.

BIOMASS DATA COLLECTION

Estimates of biomass production will be obtained through the use of clip plots. Usually 0.25-m^2 or 0.5-m^2 quadrats are used for clip plots. The specific size and how to randomly locate the quadrats in the sample area will be provided in the SAP. A list of all species found rooted within the quadrat is recorded on the datasheet. All of the current years' growth of herbaceous species is clipped at ground level (<1 cm), sorted by species, placed into individual paper bags, and labeled with the species and sample number. The previous years' growth is considered litter, even though it may still be standing erect. No cacti or woody vegetation are used to obtain biomass data. If needed, a list of species not to be included (i.e., semiwoody species) can be developed to ensure the consistency of the biomass collection. If specified in the SAP, the plant litter from each quadrat can also be collected and placed into individually labeled paper bags. Bags should be stored in a dry, cool location.

In the laboratory, a drying curve should be established to determine the length of drying time (at 65°C) necessary to obtain constant weights (i.e., so no further water loss occurs). The clipped materials (still in the paper bags) should be oven dried at 65°C, and then the weight of the clipped vegetation should be determined. The total bag weight minus the empty bag weight equals the plant biomass. Weights should be determined to the nearest 0.1 gram and recorded on the datasheet.

PLANT TISSUE DATA COLLECTION

Collection of plant material for tissue analysis is conducted like that for biomass data collection with some notable differences. Generally, specific plant species are collected for tissue analysis. The list of species to be collected and the plant organs to be collected (i.e., roots, stems, flowers, fruits, seeds, or entire plant), along with the amounts of each type of material needed for analysis, will be specified in the SAP. This SOP assumes that only aboveground biomass is to be collected.

Co-location of biomass and plant tissue for sampling is important for interpretation of results. Species collected for tissue analysis should come from the area surrounding the quadrat used for biomass sampling. Plant tissue of the species of interest is clipped off at ground level (< 1cm) in concentric circles of increasing size around the biomass quadrat. If the SAP specifies that only seeds are to be collected, then the whole plant need not be clipped, but seeds would be collected from increasing concentric circles around the biomass quadrat. Seeds are generally collected by hand. However, plastic sheets may be spread under mature seed bearing plants, which are then beaten, causing the seeds to fall on the plastic sheeting, where they may easily be collected. Collection of material continues in concentric circles of increasing distance from the biomass quadrat until sufficient material for analysis is collected. Collected material is placed into labeled, appropriate bags or containers as specified in the SAP. It may be necessary to use specific types of gloves or to have special types of containers for storing the plant tissues, depending on what is being analyzed for. These details should be specified in the SAP. Collected material should be stored in ice chests with re-freezable ice packs in the field until it can be transferred to more permanent cooler storage at the field laboratory. All samples will be kept at 4°C until sent to the laboratory..

REFERENCES AND OTHER SOURCES

Bonham, C.D. 1989. Measurements for terrestrial vegetation. John Wiley & Sons, New York, NY.

Brower, J.E., and J.H. Zar. 1977. Field and laboratory methods for general ecology. Wm. C. Brown Publishers, Dubuque, IA.

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SOP SD-05
SURFACE SEDIMENT SAMPLING USING AN EKMAN GRAB SAMPLER

This SOP describes the procedures used to collect surface sediment with an Ekman grab sampler. For the purposes of this SOP, surface sediment is defined as the upper 10 cm of the sediment column, but may vary given the sampling interval specified in the study design. The specific sampling interval will be specified in the project-specific field sampling plan (FSP). Surface sediment is typically analyzed for various physical and chemical variables so the sample equipment and procedure must be compatible with all analyses.

A stainless-steel Ekman grab sampler is a light-weight sampler capable of collecting acceptable samples from a variety of soft substrates, such as silt, silt mixed with clay, and silt mixed with a little sand (U.S. EPA 2001). This sampler is effective in various environments (e.g., lakes, rivers, and streams). A quiescent environment is needed for optimal sampling in deeper water. In very soft sediment it can penetrate as much as 15 cm, but in most sediment it penetrates less than 10 cm unless auxiliary weights or driverods are used. The Ekman grab sampler has two doors on top to allow easy access to the sediment for visual characterization and removal of undisturbed surface sediments. The procedures for collecting surface sediment samples using the Ekman grab sampler are described below.

EQUIPMENT AND SUPPLIES REQUIRED

Equipment required for sediment sampling using the Ekman grab sampler includes the following:

- Stainless-steel Ekman grab sampler (typically 0.25 ft^2), and spare parts
- Rope of sufficient length to reach the sediment at the deepest station plus 10 ft, and of a diameter that will fit through the messenger (<0.25 in. OD)
- Messenger (it is preferable to have two messengers)
- Metal driverods for harder sediment in shallow water
- Weights that attach to the outside of the Ekman
- Teflon® or polyethylene siphon
- Flat-bottomed container (e.g., dish pan)

- Stainless-steel ruler
- Stainless-steel spoons
- Stainless-steel mixing bowl or pot.

DECONTAMINATION

To prevent potential cross-contamination of samples, all reusable sediment sampling equipment will be decontaminated. Before each station is sampled, decontaminate the inner surfaces of the grab sampler and all stainless-steel sample compositing equipment. Details on correct decontamination procedures can be found in SOP SD-01, *Decontamination of Equipment—Sediment*. The project-specific FSP should also be consulted to determine any project-specific decontamination procedures. The personnel performing the decontamination procedures will wear protective clothing as specified in the site-specific health and safety plan.

All solvent rinsates (if used) will be collected into a bucket or tub and allowed to evaporate over the course of the day. Any rinsate that has not evaporated by the end of the sampling event will be containerized and disposed of in accordance with applicable regulations.

GRAB SAMPLER DEPLOYMENT

1. If the water depth is less than 9 ft and the sediment is relatively hard, attach the grab sampler to the metal driverods. If the water depth is greater than 9 ft, use the rope to deploy the grab sampler.
2. Place the decontaminated grab sampler on a clean surface and open it.
3. Ensure that the two release wires are securely placed around the release pins.
4. Lower the sampler through the water column at a slow and steady speed to reduce turbulence ahead of the sampler.
5. Allow the grab sampler to contact the bottom gently. In soft sediments allow only its weight to force it into the sediments. The sampler should never be allowed to "free fall" to the bottom because this may result in an excessive wake, or improper orientation upon contact with the bottom.
6. Release the messenger to close the doors on the bottom of the grab sampler.

GRAB RETRIEVAL

1. After the grab sampler has rested on the bottom for approximately 5 seconds, begin retrieving it at a slow and steady rate. This will avoid disturbing the sediment interface and loss of sediment from tilting or washout during retrieval.

2. After the grab sampler breaks the water surface, gently lower it into a clean, flat-bottomed container, while maintaining the grab sampler in an upright position. This will help retain an intact interface by reducing how much the overlying water sloshes back and forth. If the jaws are not completely closed, push them closed by hand.

3. As soon as the grab sampler is secured, open the doors on the top of the grab sampler, and inspect the sample for acceptability. The following acceptability criteria should be satisfied:

   - The sampler is not overfilled with sample to the point that the sediment surface presses against the top of the sampler or is extruded through the top of the sampler

   - Overlying water is present (indicating minimal leakage)

   - The overlying water is not excessively turbid (indicating minimal disturbance or winnowing)

   - The sediment surface is relatively undisturbed; the sediment-water interface is intact and relatively flat with no sign of channelling or sample washout

   - The desired penetration depth is achieved

   - There is no sign of sediment loss (incomplete closure of the sampler, penetration at an angle, or tilting upon retrieval).

If a sample fails to meet the above criteria, it will be rejected and discarded away from the station. The locations of replicate samples should be as close to the first sample as possible, and if sampling on a river or stream, samples should be located upstream of previous samples. Rejected sediment samples should be discarded in a manner that will not affect subsequent samples at that station or other possible sampling stations.

Penetration depth should be determined with a decontaminated stainless-steel ruler by measuring the distance from the top of the sampler to the sediment interface and subtracting this from the inside depth of the sampler. If the sample is fairly level inside the sampler, this measurement can be made at one edge. If the sample is uneven but has an intact interface, then measurements should be made on opposite edges of the sample and the average value used. This observation (i.e., that the sediment surface is slanted) and subsequent calculation of the average penetration depth should be recorded in the field logbook. If penetration depth is inadequate, add auxiliary weights. If penetration is still too shallow, a Ponar or van Veen grab sampler may be necessary.

SAMPLE REMOVAL AND PROCESSING

1. For acceptable samples, remove the overlying water by slowly siphoning it off near one or more sides of the grab sampler. Ensure that the siphon does not contact the sediments or that fine-grained sediment is not entrained and siphoned off. If sediment is suspended in the overlying water, do not proceed with siphoning until the sediment is allowed sufficient time to settle.

2. After the overlying water is removed, characterize the sample as specified in the study design. Characteristics that are often recorded include:

   - Sediment type (e.g., silt, sand)
   - Texture (e.g., fine-grain, coarse, poorly sorted sand)
   - Color
   - Presence/location/thickness of the redox potential boundaries (a visual indication of black is often adequate for documenting anoxia)
   - Approximate percentage of water
   - Biological structures (e.g., chironomids, tubes, macrophytes)
   - Approximate percentage of biological structures
   - Presence of debris (e.g., twigs, leaves)
   - Approximate percentage of organic debris
   - Presence of shells
   - Approximate percentage of shells
   - Stratification, if any
   - Presence of a sheen
   - Odor (e.g., hydrogen sulfide, oil, creosote).

3. After the sample is characterized, remove the top 10 cm using a stainless-steel spoon (see project-specific FSP for correct sampling interval) and place in a decontaminated stainless steel bowl. Unrepresentative material (e.g., large shells, stones, leaves, twigs) should be carefully removed without touching the sediment sample under the supervision of the field team leader and noted in the field logbook.

4. Remove subsamples for analysis of unstable constituents (e.g., volatile organic compounds, acid-volatile sulfides), and place them directly into sample containers without homogenization. Sediment must be placed in these containers with no headspace and no entrapped bubbles (i.e., completely fill the sample container).

5. Transfer the remaining surface sediment to a stainless-steel mixing bowl or pot for homogenization. Additional grab samples may be required to collect the volume of sediment specified in the study design. If it is necessary to collect additional grab samples to meet the project-specific volume requirements, the mixing bowl or pot should be covered with aluminum foil (dull side down) to prevent sample contamination (e.g., from precipitation, splashing water, falling leaves) and placed out of the sun and away from heat.

6. After the surface sediment has been removed from the grab sampler, move away from the station, open the jaws of the grab sampler, and allow the remainder of the sediment to fall out of the grab sampler. Discard this material away from the station, and rinse away any sediment adhering to the inside of the grab sampler. This can be done by repeatedly dipping the sampler in the water. The grab sampler is now ready for additional sampling at the same station or decontamination before sampling at a new station.

7. After a sufficient volume of sediment is transferred to the mixing bowl, homogenize the contents of the bowl using stainless-steel spoons until the texture and color of the sediment appear to be uniform.

8. After the sample is homogenized, distribute subsamples to the various containers specified in the study design and preserve the samples as specified in the study design.

9. After all sediment for testing has been placed in the sample containers, if it is suspected that there is a clay component to the sediment, a "ribbon test" should be performed on the sediment to confirm this suspicion. In this "texture-by-feel" test, a small piece of suspected clay is rolled between the fingers while wearing protective gloves. If the piece easily rolls into a ribbon it is clay; if it breaks apart, it is silt. This information should be noted in the field logbook.

FIELD QUALITY CONTROL SAMPLES

Details on collection of field quality control samples and preparation of the certified reference materials can be found in SOP SD-02, *Preparation of Field Quality Control Samples—Sediment* and SOP SD-03, *Preparation of Reference Materials—Sediment.*

FIELD MEASUREMENTS

A water depth measurement must be collected at every sampling location. For sites where tides affect water depth, the time of collection and depth measurement must also be recorded. Depending on the specific project objectives, it may be necessary to perform field measurements of the *in situ* environment. Possible field measurements include temperature and pH of the sediment at the sediment-water interface and concentration of dissolved oxygen, salinity or conductivity in the overlying water. Details on collection of field measurements can be found in SOP SD-19, *Field Measurements for Sediment.* Required field measurements, if any, will be specified in the project-specific FSP.

STATION LOCATION COORDINATES

Station locations will be determined in accordance with the projects field sampling plan. Generally, station locations are determined with a differential global positioning system (DGPS), which is capable of providing latitude and longitude coordinates with an accuracy of approximately 2 m. The DGPS consists of two satellite receivers linked to each other by a VHF telemetry radio system. The GPS receiver will be on the sampling vessel. Details on collection of field station coordinates can be found in SOP GEN-04, *Station Positioning Using the Trimble Pathfinder Pro XRS.*

REFERENCE

U.S. EPA. 2001. Methods for collection, storage and manipulation of sediments for chemical and toxicological analyses: Technical Manual. EPA-823-B-01-002. U.S. Environmental Protection Agency, Office of Water, Office of Science and Technology, Washington, DC.

Exponent

SOP SD-10
SEDIMENT CORING USING A DRIVE ROD CHECK VALVE CORER

This standard operating procedure (SOP) describes the procedure for collecting sediment core samples using a drive rod check valve corer. The drive rod check valve corer is designed for collecting short cores (<60 cm) in water less than about 30 ft deep. The corer is lowered through the water column and then driven into the sediment using drive rods. This corer has the advantage over gravity corers in that the drive rods allow up to 200 lb of driving force to be used without having to handle or lift a heavy weight.

The sample is held in the core tube with the suction provided by a check valve at the top of the corer. Unlike free-floating check valves, this valve is actuated from the boat using a cord. As the corer is lowered, the valve is held open so water flows freely through the corer as it approaches the sediment, thus reducing the wake that can disrupt the surficial sediments. Because it is not a piston-type corer, some bypass/compaction of the sample will occur depending on the sediment type and core length. The internal cross-sectional area of the 3-in. diameter corer is 39 cm^2, which yields about 2 g of dry solids per centimeter of sample thickness at a porosity of 98 percent and about 15 g of solids per centimeter of thickness at a porosity of 85 percent.

There are five basic steps to collecting sediment with this corer:

1. Prepare the corer
2. Measure the water depth
3. Drive the corer
4. Retrieve the corer
5. Remove the core.

When reading instructions, refer to Figures SD-10-1 through SD-10-4.

PRELIMINARY CONSIDERATIONS

It is best to work from a platform that is anchored and will not drift. This setup helps to prevent collecting a poor quality sample and damaging the equipment. A platform with a low free-board, such as a pontoon boat, is best.

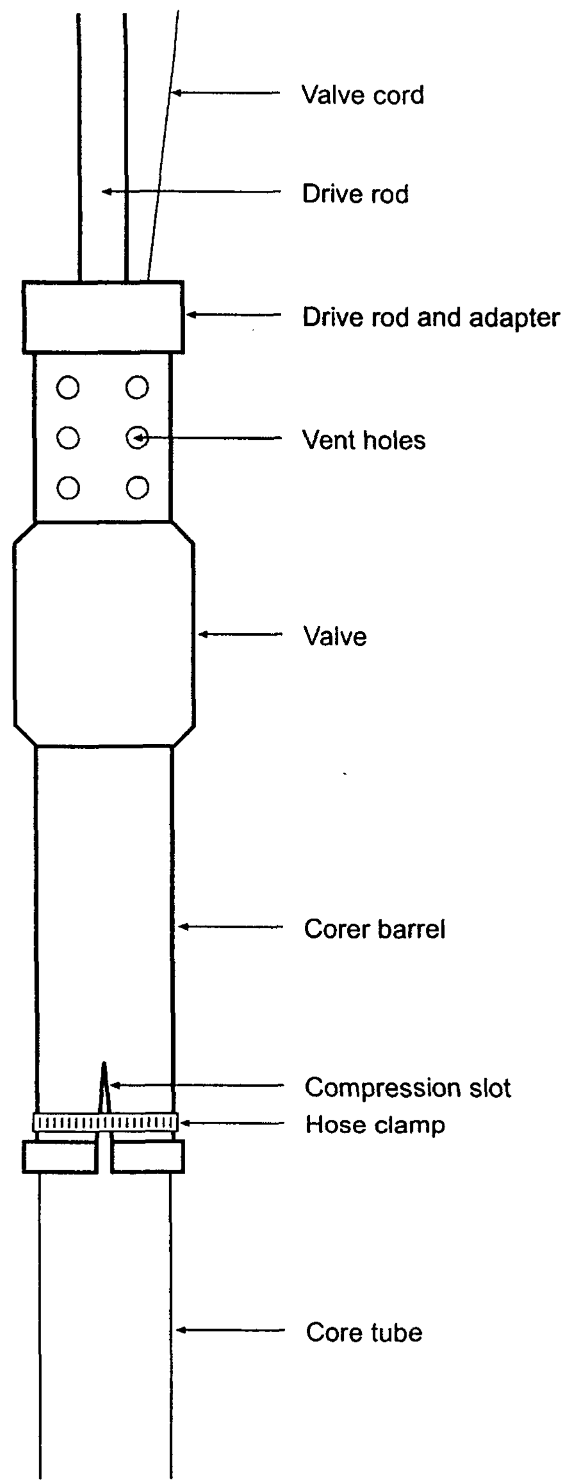

Figure SD-10-1. Drive rod check valve corer.

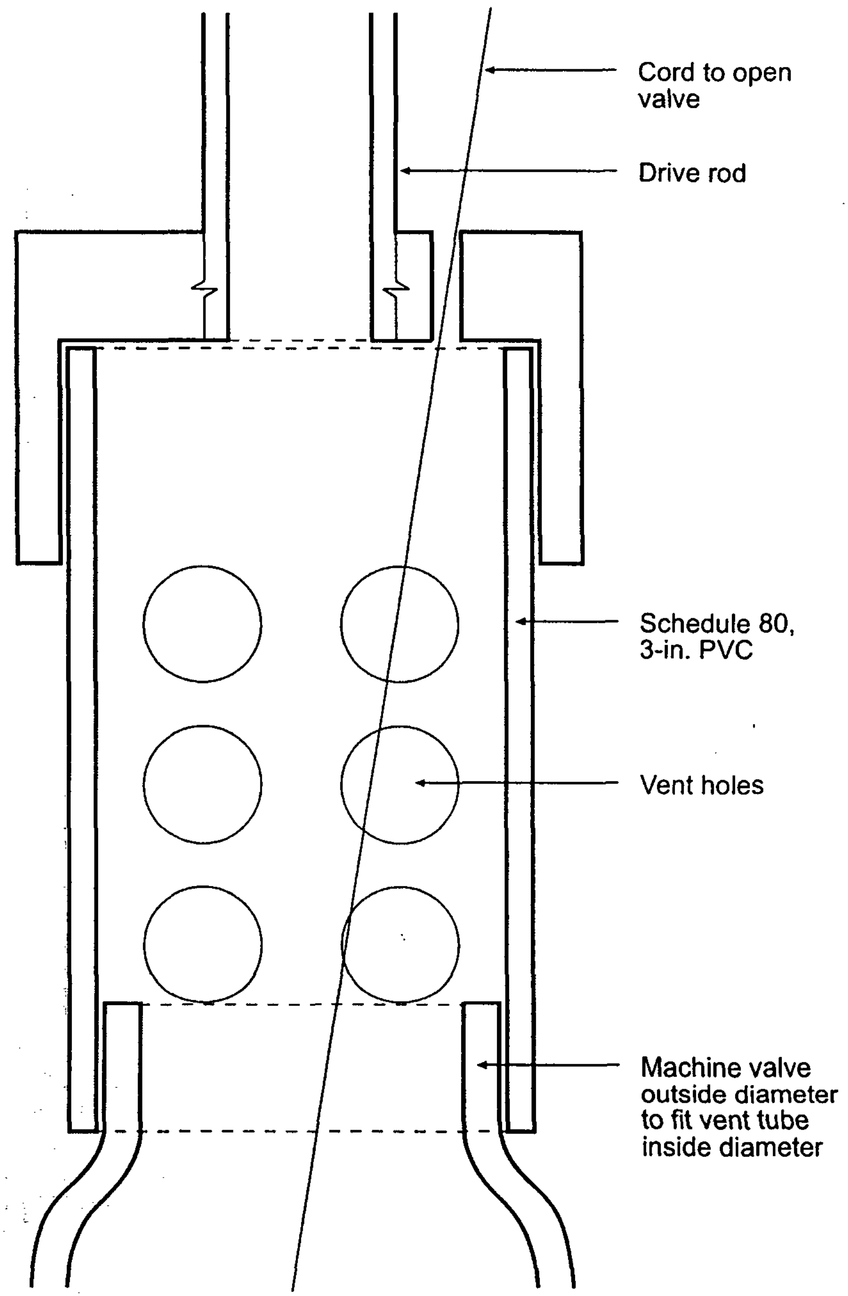

Figure SD-10-2. Detail of vent tube.

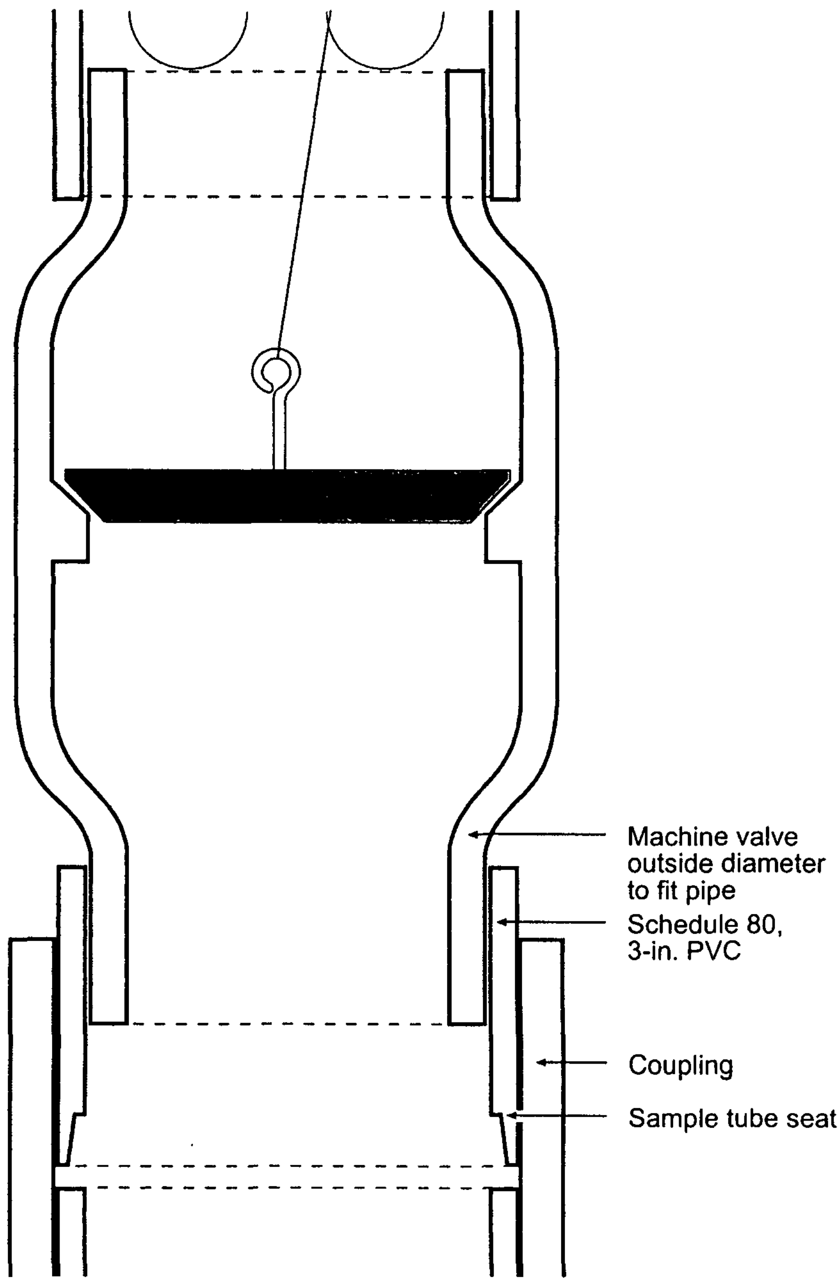

Figure SD-10-3. Detail of valve.

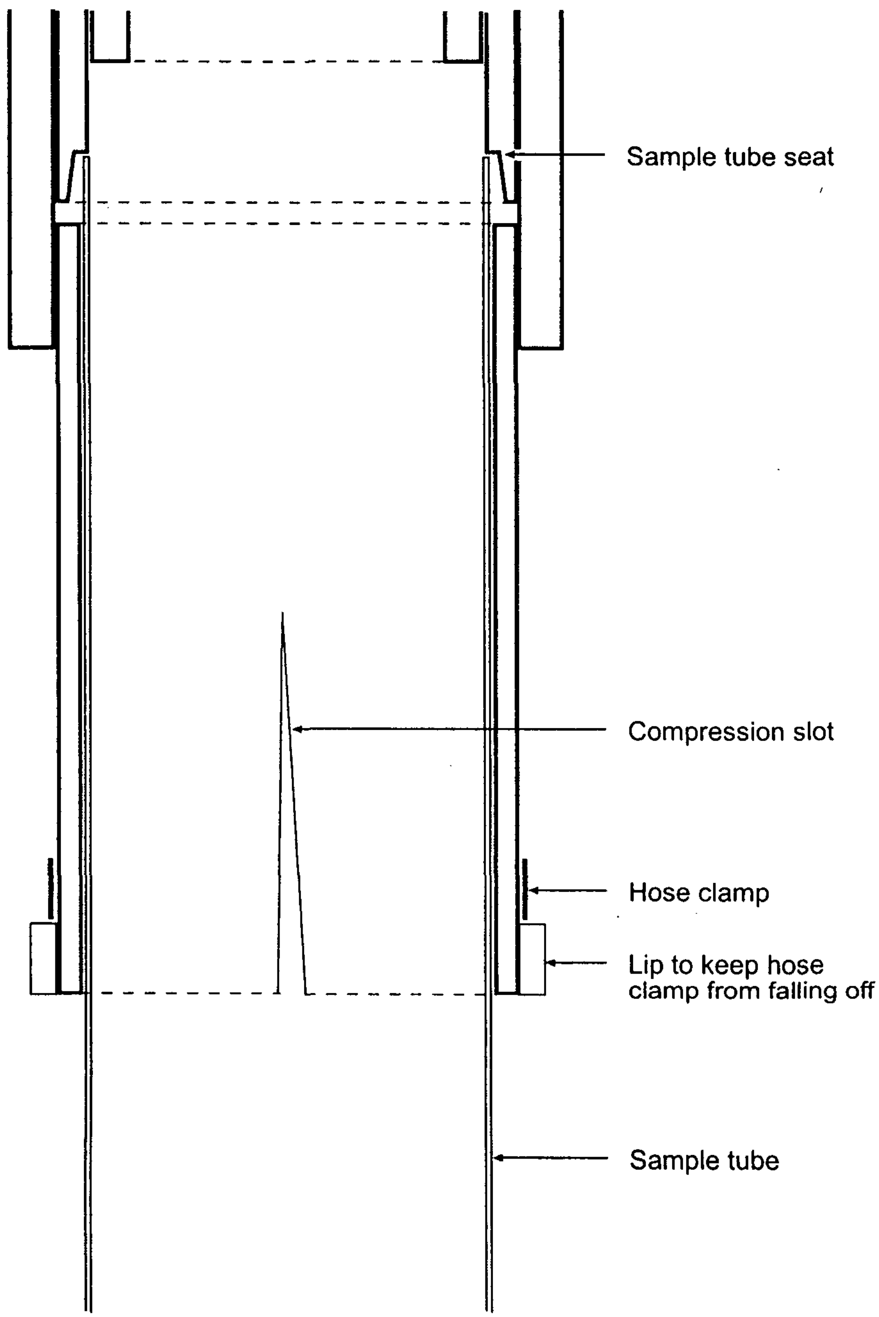

Figure SD-10-4. Detail of sample tube holder.

Core tubes can vary in length from about 70 to 200 cm. The core tube should be about 50 cm longer than the sample length needed to provide for overlying water and errors in the depth driven. It is desirable to have about 20 to 30 cm of water overlying the sediment in the core tube. The overlying water provides a buffer that reduces agitation of the surfical sediments when handling the core tube.

The corer should be pushed into the sediments deeper than the length of core needed. If the sediments are soft, it is possible to over penetrate and run the sediment–water interface up into the valve. A long core tube will help prevent such an occurrence. For the tube to retain the sample, the minimum sample length is about 3 to 4 times the diameter depending on the sediment type.

DECONTAMINATION

To prevent potential cross-contamination of samples, all reusable sediment sampling equipment will be decontaminated. Before each station is sampled, decontaminate the inner surfaces of the corer or core tube liner and all stainless-steel sample compositing equipment. Details on correct decontamination procedures can be found in SOP SD-01, *Decontamination of Equipment—Sediments.* The project-specific field sampling plan (FSP) should also be consulted to determine any project-specific decontamination procedures. The personnel performing the decontamination procedures will wear protective clothing as specified in the site-specific health and safety plan.

All solvent rinsates (if used) will be collected into a bucket or tub and allowed to evaporate over the course of the day. Any rinsate that has not evaporated by the end of the sampling event will be containerized and disposed of in accordance with federal regulations.

INSTRUCTIONS

1. **Prepare the Corer**—Before using the corer, inspect it for worn or broken parts, and repair as necessary.

   1.1 Clean the corer; sandy material in particular can foul the valve and other seals. If the corer has been used in a sandy area, sand caught on the seat might prevent the valve from sealing. To clean the valve, run or spray water through it while repeatedly opening and closing the valve. Test the valve for leaks by releasing the valve cord and pouring water into the top of the corer and watching for leakage. No more than about 1 mL per minute should leak.

   1.2 Insert a core tube into the corer barrel and push it in until you feel the top end of the tube contact the sealing ring at the top of the corer barrel. To seat the tube, push it hard for about a tenth of an inch; you will feel it seat into position. If the bottom edge of the core tube is beveled to improve cutting action, make sure the tube is not upside down. Tighten the hose clamp at the bottom of the barrel so that the core tube cannot be rotated by hand within the corer barrel. Make sure that the drive rod is tightly screwed into the adapter.

2. **Measure the Water Depth**—Measure the water depth to within about a foot of the true depth, using a weighted measuring tape or sonar.

   You will need to know the depth so you can attach the correct length drive rods and so you can determine how close the corer is to the sediment as it is being lowered.

3. **Drive the Corer**

   3.1 While keeping the valve open with the valve line, lower the corer and keep adding drive rods until the corer is near the sediment. Only a couple pounds of lifting force is required to keep the valve open, so do not lift too hard on the valve line. With the corer and drive rods hanging vertically, lower the corer slowly until you feel it contact the sediment, and then with one smooth motion, push the corer into the sediment. Be careful to push vertically on the corer. If the platform moves laterally and the drive rods are at an angle, attempting to drive the corer may damage it.

   3.2 After the corer is driven to the desired depth, release the valve cord so the valve closes.

4. **Retrieve the Corer**—After the valve is closed, the corer can be retrieved; retrieval is best done with two people.

   4.1 Lift steadily on the drive rods until you feel the corer break loose from the sediments. As the corer approaches the water surface, have a rubber stopper ready to place in the bottom of the core tube. If the sediments are sandy and the samples tend to erode from the bottom of the tube as it is lifted through the water column, it may be necessary to keep the corer submerged just below the surface while another person reaches underwater and places the stopper in the tube. If sampling is performed from a large boat that has a lot of free-board, it may be necessary to have someone near the water level on a skiff to insert the stopper. While the corer is being lifted onboard, support the rubber stopper so that it and the sample do not fall out.

   4.2 After the corer is onboard, seat the stopper so it is entirely inside the core tube by placing a second stopper on the deck and pushing the corer down on top of it. Keep the corer vertical at all times to prevent the sample from shifting, and avoid rapid movements that can disrupt the interface.

5. **Remove the Core**

   5.1 As a second person holds the corer vertical and keeps the valve open, loosen the hose clamp at the bottom of the core barrel and hold the bottom of the core tube firmly against the deck.

   5.2 While holding the core tube, have the second person lift the corer off the tube. If the tube is seated very firmly in the barrel from the force of driving the corer, twist the barrel slightly while lifting it off the tube to break it loose. It is best to rotate the barrel, not the core tube, because when it breaks loose, the rapid rotation of the core tube may disrupt the sediment–water interface. As the core barrel is lifted off the tube, the water in the valve assembly will spill. Before moving or lifting the core tube, seal the top of the core tube with a test plug.

   5.3 Inspect the sample for acceptability. The following acceptability criteria should be satisfied:

   - The sampler is not overfilled with sample so that the sediment surface presses against the top of the sampler or is extruded through the top of the sampler
   - Overlying water is present
   - The overlying water is not excessively turbid (indicates minimal disturbance)
   - The desired penetration depth is achieved (see project-specific FSP for required penetration depth)

   5.4 If possible, extrude and section the sample immediately in accordance with SOP SD-08, *Sediment Coring Procedures Using Slide-Hammer and Gravity Corers*. Immediate extrusion and sectioning is essential if the sample is to be analyzed for redox-sensitive elements. Oxygen diffuses through the polycarbonate core tube and oxidizes ferrous iron in the pore water. This process is fairly rapid, and an orange iron oxide precipitate will visibly form on the inside walls of the core tube within a day. There is some evidence that this oxidation does not extend more than a couple millimeters into the sample. If the sample cannot be extruded immediately, keep it cool and out of the sun by refrigerating it or wrapping it with aluminum foil.

SAMPLE REMOVAL AND PROCESSING

1. For acceptable samples, remove the overlying water by slowly siphoning it off near one or more sides of the corer. Ensure that the siphon does not contact the sediments or that fine-grained suspended sediment is not siphoned

off. If sediment is suspended in the overlying water, do not proceed with siphoning until the sediment is allowed sufficient time to settle.

2. After the overlying water is removed, characterize each sampling interval as specified in the study design. Characteristics that are often recorded in the field logbook include:

   – Sediment type (e.g., silt, sand)

   – Texture (e.g., fine-grain, coarse, poorly sorted sand)

   – Color

   – Presence/location/thickness of the redox potential discontinuity layer (a visual indication of black is often adequate for documenting anoxia)

   – Approximate percentage of water

   – Presence of biological structures (e.g., chironomids, tubes, macrophytes)

   – Presence and approximate percentage of biological structures, organic debris

   – Stratification, if any

   – Presence of a sheen

   – Odor (e.g., hydrogen sulfide, oil, creosote).

3. After the sample is characterized, remove the specified sampling interval using a stainless-steel spatula or spoon (see project-specific FSP for correct sampling interval). Unrepresentative material (e.g., large shells, stones) should be carefully removed without touching the sediment sample under the supervision of the field team leader and noted in the field logbook.

4. Remove subsamples for analysis of unstable constituents (e.g., volatile organic compounds, acid-volatile sulfides), and place them directly into sample containers without homogenization. Sediment must be placed in these containers with no headspace and no entrapped bubbles (i.e., completely fill the sample container).

5. Transfer the remaining surface sediment to a stainless-steel mixing bowl or pot for homogenization. Additional sediment may be required to collect the volume of sediment specified in the project-specific FSP. If it is necessary to collect multiple cores at a station to meet the project-specific volume requirements, then the mixing bowl or pot should be covered with aluminum foil (dull side down) to prevent sample contamination (e.g., from precipitation, splashing water).

6. After removing the sediment from the corer, move the sampling vessel away from the station and discard this material away from the station, and rinse away any sediment adhering to the inside of the corer. The corer is now ready for additional sampling at the same station or decontamination before sampling at a new station.

7. After a sufficient volume of sediment is transferred to the mixing bowl or pot, homogenize the contents of the bowl or pot using stainless-steel spoons until the texture and color of the sediment appears to be uniform.

8. After the sample is homogenized, distribute subsamples to the various containers specified in the study design and preserve the samples as specified in the study design. The sediment in the mixing bowl or pot should be briefly stirred in between each spoon transfer to the sample containers.

9. After all sediment for testing has been placed in the sample containers, if it is suspected that there is a clay component to the layer of sediment, a "ribbon test" should be performed on the sediment collected from this layer to confirm this suspicion. In this "texture-by-feel" test, a small piece of suspected clay is rolled between the fingers while wearing protective gloves. If the piece easily rolls into a ribbon it is clay; if it breaks apart, it is silt. This information should be noted in the field logbook.

FIELD QUALITY CONTROL SAMPLES

Details on collection of field quality control samples (e.g., field duplicates) will be specified in the project-specific FSP. Details on collection of field quality control samples and preparation of the certified reference materials can be found in SOP SD-02, *Preparation of Field Quality Control Samples—Sediment* and SOP SD-03, *Preparation of Reference Materials—Sediment*. Not all of the field quality control samples discussed in this SOP may be required for a given project. The specific field quality control samples will be described in the project-specific FSP and quality assurance project plan (QAPP).

FIELD MEASUREMENTS

A water depth measurement must be collected at every sampling location. Depending on the specific project objectives, it may be necessary to perform field measurements of the *in situ* environment. Possible field measurements include temperature and pH of the sediment at the sediment-water interface and concentration of dissolved oxygen, salinity, or conductivity in the overlying water. Details on collection of field measurements can be found in SOP SD-19, *Field Measurements for Sediment*. The specific field measurements, if any, will be specified in the project-specific FSP.

STATION LOCATION COORDINATES

At a minimum, station locations for all field sampling will be determined using a differential global positioning system (DGPS), which is capable of providing latitude and longitude coordinates with an accuracy of approximately 3 m. More accurate determination of position may be required by the project FSP. Details on collection of field station coordinates can be found in SOP GEN-04, *Station Positioning Using the Trimble Pathfinder Pro XRS*.

SAMPLE CUSTODY AND SAMPLE SHIPPING

Sample custody will be maintained in accordance with procedures outlined in SOP GEN-02, *Sample Custody*. All samples will be packaged and shipped with other samples in accordance with procedures outlined in SOP GEN-03, *Sample Packaging and Shipping*.

TROUBLESHOOTING

Problem 1: The Corer Is Not Retaining the Sample

There are two possible causes for this problem. One is that the sediments are sandy and not cohesive so they do not stick to the core tube walls or themselves. As a result, the core erodes from the bottom as it is lifted through the water. This problem can be solved in several ways.

- Drive the corer deeper into the sediments, where there may be a more cohesive layer. It is not unusual for a fine-grained cohesive layer to lie below coarser layers.
- Place a stopper in the bottom of the tube as soon as possible using one of two methods: 1) use a rod that holds the stopper in the correct position, maneuver the rod below the tube, and lift it up to insert the stopper, or 2) have a diver insert the stopper.
- Use a smaller diameter corer so there is relatively more cohesion of the sediment with the walls.

The second possible cause is a leak in the suction of the corer that allows the whole core to start slipping out of the core tube. There are two places where the suction can be lost: the valve, and the seat between the core barrel and the core tube. Inspect and clean both the valve and the seat, and check to be sure that the valve is not stuck in the open position.

Problem 2: The Sediment Interface Is Not Distinct

There are several possible causes for this problem. One is that the bottom end of the core tube was moving horizontally when it first contacted the sediments. Further evidence of this cause is if the sediment interface is tilted. In this case, make sure the platform is not moving and that the corer and drive rods are allowed to hang vertically just before driving the corer. Another common cause is the formation of gas bubbles in the sediments of productive or eutrophic systems. When a corer is pushed into this type of sediment, bubbles are released that entrain and resuspend sediment. There is no easy solution to this problem other than to let the resuspended sediment settle before processing the sample. Another possible cause is rough handling of the sample.

Problem 3: The Core Recovery Is Low

Little can be done to prevent bypass/compaction other than to use a piston corer. However, the amount of bypass/compaction can be quantified. One easy method is to apply Velcro® tape to the outside of the corer barrel and determine the depth of penetration by noting where sediment is caught in the Velcro®.

APPENDIX B

AEC QA/QC MANUAL
(Available upon request, included in RI SAP)